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COVER: ***Andrena* sp. bee (Hymenoptera: Andrenidae)**

A male *Andrena* sp. bee (Hymenoptera: Andrenidae) foraging on sage buttercup (*Ranunculus glaberrimus*) in the central Okanagan, 11 March 2015. In this issue Sheffield and Heron present a checklist of the bees of British Columbia which includes 483 species – 37 of which are new provincial records and 20 of which are new Canadian records.

Photograph details:
Photograph by Robert Lalonde (UBC Okanagan). This photograph was made with a Canon EOS digital rebel T2i equipped with a Canon 100mm macro lens; ISO 800; f5.6 at 1/320 sec.

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First records of *Baetis vernus* Curtis (Ephemeroptera: Baetidae) in North America, with morphological notes

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ABSTRACT

The *Baetis vernus* group (Ephemeroptera: Baetidae) – which includes *B. brunneicolor* McDunnough, *B. bundyae* Lehmkuhl, *B. hudsonicus* Ide, *B. jaervii* Savolainen, *B. liebenauae* Keffermüller, *B. macani* Kimmins, *B. subalpinus* Bengtsson, *B. tracheatus* Keffermüller & Machel, and *B. vernus* Curtis – is both diverse and taxonomically tangled. Some members of the group – *B. brunneicolor*, *B. bundyae*, and *B. hudsonicus* – have been previously found in North America. The remainder of the group is known to be only of Palearctic distribution, including *B. vernus,* which has a wide trans-Palearctic distribution. We report the collection of specimens from the Northwest Territories and British Columbia that we have identified as *B. vernus* using DNA barcoding and morphological examination and provide characters to assist separation of the North American members of the group from *B. vernus*. A genetically cohesive Holarctic clade for *B. vernus* likely relates to a Beringian dispersal event. This substantial expansion of the known range of *B. vernus* adds new phylogeographic and ecological complexity, but it may also help to provide further clues to the evolutionary history of this group.

INTRODUCTION

Mayflies of the *Baetis vernus* group (Savolainen *et al.* 2007; Ståhls and Savolainen 2008; Drotz *et al.* 2012) are widespread across the Holarctic, but distributions of its members are challenging to determine because many are difficult to separate in the most commonly collected larval stage using morphological characters (Ståhls and Savolainen 2008; Drotz *et al.* 2012). This is due both to similarity of characters among group members and to high levels of variation relating to environmental conditions (Bauernfeind and Humpesch 2001; Ståhls and Savolainen 2008). Ståhls and Savolainen (2008) stressed the importance of combining molecular and morphological data to sort out species distributions in this group.

Until recently, only three species in this group were known in North America (McCafferty and Jacobus 2017). *Baetis brunneicolor* McDunnough is widespread in the Nearctic: it is reported from across Canada, including Arctic and Sub-Arctic zones (Harper and Harper 1981; Cordero *et al.* 2017; Giberson and Burian 2017) and is found in the northeastern, northwestern, and southeastern United States (USA; McCafferty and Jacobus 2017). *Baetis bundyae* Lehmkuhl has a generally northern distribution in Nearctic and Palearctic: in North America, it is widespread across the north but also extends into the northern USA (Giberson *et al.* 2007; Giberson and Burian 2017). *Baetis hudsonicus* Ide has so far been reported only in northern and far northern Canada (Cordero *et al.* 2017; Giberson and Burian 2017; McCafferty and Jacobus 2017).

Recent collecting efforts in northern British Columbia (Huber *et al.* 2019) and in the Northwest Territories (Cordero *et al.* 2017) revealed four specimens whose cytochrome

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oxidase I (COI) barcode matched Palearctic specimens of *Baetis vernus* Curtis, a species not previously reported in North America. *Baetis vernus* specimens showed many morphological similarities to *B. brunneicolor*, potentially causing confusion when determining the distribution of the two species in North America. Recently, Webb *et al.* (2018) recommended that *B. brunneicolor* and *B. vernus* be treated as a species complex (the *B. vernus* complex) and not identified further if identifying larvae using current morphologically-based keys. Here, we describe DNA barcode data of the Canadian *B. vernus* specimens, demonstrating their genetic similarity to Palearctic *B. vernus* and distance from other *B. vernus* group members, as well as the relevant morphology of those same specimens compared to North American *B. brunneicolor* characteristics.

METHODS AND MATERIALS

Mayfly larvae examined in this study resulted from recent aquatic insect sampling in river and lake habitats in northern British Columbia (BC), Yukon (YT) and Northwest Territories (NT), plus examination of archived specimens in the Canadian National Collection (CNCI) in Ottawa (Giberson and Burian 2017; Huber *et al.* 2019). Collection locality, voucher, and DNA sequence data for specimens that were collected and/or analyzed in this study are described in Table 1. The cytochrome oxidase I (COI) barcode region (Hebert *et al.* 2003; Ball *et al.* 2005) of the Crooked River, BC, specimen was sequenced at the Biodiversity Institute of Ontario, and other barcode sequence data were extracted from public databases. North American *B. vernus* group specimens were compared to other described members of the *B. vernus* for which sequence data were available (exceptions: *B. jaervii* Savolainen and *B. tracheatus* Keffermüller & Machel). A FASTA file of all sequences used in this study, including sequence ID and accession information, is available as supplemental data. All sequence data are publicly available as listed in Table 1 and as BOLD IDs (most sequences) or an NCBI accession number (Yellowknife specimen sequence) in Figure 1. Barcode sequences were aligned with ClustalW and visualized with FigTree v.1.4.3.

Specimens were observed for morphological characters, colouration, and colour patterns under Wild M5A stereoscopic and Bausch & Lomb phase contrast compound light microscopes (up to 1000x magnification). Mouth and body parts of the larvae were dissected in 80% alcohol and slide mounted in Hoyer's Mounting Media. Specimens were photographed using a Nikon D300s DSLR and the Nikon Camera Control Pro2® software. All measurements were made using a calibrated ocular micrometer (nearest 0.10 mm). Measurements were made from entire specimens and/or parts (not mounted on slides) that were held as flat as possible (without inducing distortion) using sections of broken glass microscope slides and coverslips.

Specimens were determined to species by comparing morphological characters to all pertinent descriptions and morphological studies of members of the *Baetis vernus* species group on a global basis, as well as Nearctic keys to species of *Baetis* (Ide 1937; Leonard 1950; Macan 1957; Keffermüller and Machel 1967; Müller-Liebenau 1969; Lehmkuhl 1973; Morihara and McCafferty 1979a, b; Jacob 2003; Wiersema *et al.* 2004; Savolainen 2009; Jacobus *et al.* 2014). In addition, reared specimens of *B. brunneicolor* from the USA and voucher specimens of larvae of *B. macani* and *B. jaeverii* (provided by E. Savolainen) from Finland were used to evaluate characters observed on *B. vernus* specimens from northern Canada.

Table 1

Collection data for *Baetis* specimens in this study. All specimens listed were verified to species by S.K. Burian, except BIOUG22893-F05 and BIOUG22893-F05 (verified by J.E. Sones) and the specimens listed in Harper and Harper (1981) (verified by P. Harper) and Cordero *et al.* 2017 (verified through barcoding). BC: British Columbia; NT: Northwest Territories; YT: Yukon Territory; MB: Manitoba; PQ: Province of Quebec; ON: Ontario.

Specimen code	Reg.	Site	Date	Species	Location Latitude/ Longitude	Sequence ID	Voucher location	Reference
E18-CR6	BC	Crooked River	18-Jun-14	Baetis vernus Curtis	54.328, –122.669	CREPH018-16	BIO (Univ of Guelph)	Huber et al. 2019
2011215 EYK00	NT	Near Yellowknife	10-Jun-11	Baetis vernus Curtis	62.53, –114.97	KJ675352	ROM (Toronto)	Cordero et al. 2017, Giberson and Burian 2017
BIOUG22893-F05	BC	Canyon Creek	08-Jul-14	Baetis vernus Curtis	54.768, –126.936	MG383307	BIO (Univ of Guelph)	Huber et al. 2019
BIOUG22893-F06	BC	Canyon Creek	08-Jul-14	Baetis vernus Curtis	54.768, –126.936	MG376386	BIO (Univ of Guelph)	Huber et al. 2019
1083318	NT	Harris River	30-Jun-72	Baetis brunneicolor McD.	61.867, –121.317		CNCI (Ottawa)	Giberson and Burian 2017
1083321	NT	Harris River	30-Jun-72	Baetis brunneicolor McD.	61.867, –121.317		CNCI (Ottawa)	Giberson and Burian 2017
1082864	YT	Caribou Bar Creek	14-Aug-73	Baetis brunneicolor McD.	67.467, –140.550		CNCI (Ottawa)	Giberson and Burian 2017
1082878	YT	Caribou Bar Creek	28-Aug-73	Baetis brunneicolor McD.	67.467, –140.550		CNCI (Ottawa)	Giberson and Burian 2017
1082880	YT	Caribou Bar Creek	04-Sep-73	Baetis brunneicolor McD.	67.467, –140.550		CNCI (Ottawa)	Giberson and Burian 2017
1082336	NT	Bosworth Creek	03-Jul-08	Baetis brunneicolor McD.	65.302, –126.675		CNCI (Ottawa)	Giberson and Burian 2017
1082337	NT	Moosehide Creek	03-Jul-08	Baetis brunneicolor McD.	65.266, –126.736		CNCI (Ottawa)	Giberson and Burian 2017
1082563	YT	Seaforth Creek	24-Jul-06	Baetis brunneicolor McD.	60.446, –133.582		CNCI (Ottawa)	Giberson and Burian 2017
-	MB	Churchill and area (loc.22)	8 July-19 Sept 1947–1957	Baetis brunneicolor McD.	58.768, –94.165		CNCI (Ottawa)	Harper and Harper 1981
-	PQ	SEBJ basin de la riviera du castor (loc.33)	8 July-19 Sept 1973–75	Baetis brunneicolor McD.	53.4194, –78.6134		SEBJ / Collection Entomo-logique de U de M	Harper and Harper 1981
2010568 EMO043	ON	Moos-enee	19 July – 26 July 2010	Baetis brunneicolor McD.	51.2792, –80.6615	KJ675138	ROM (Toronto)	Cordero et al. 2017
2010621 ESV011	NL	Labra-dor, near border with Scheffer-ville PC	10-Jul-10	Baetis brunneicolor McD.	54.6684, –66.7636	KJ675281	ROM (Toronto)	Cordero et al. 2017
1082761	YT	Caribou Bar Creek	28-Jul-72	Baetis n.sp. (vernus group)	67.650, –140.733		CNCI (Ottawa)	Giberson and Burian 2017
1082848	YT	Caribou Bar Creek	02-Aug-73	Baetis n.sp. (vernus group)	67.533, –140.583		CNCI (Ottawa)	Giberson and Burian 2017
1082348	NT	Kat Creek	06-Aug-10	Baetis n.sp. (vernus group)	64.940, –127.457		CNCI (Ottawa)	Giberson and Burian 2017
DG 18	NT	Heart Lk/ Butterfly Ck	07-Aug-10	Baetis n.sp. (vernus group)	64.961, –127.563		NEL (NE Ephem.Lab Southern Connecti-cut State U.)	Giberson and Burian 2017

RESULTS AND DISCUSSION

DNA barcodes for *B. vernus* specimens collected by us (Cordero *et al.* 2017; Huber *et al.* 2019) and others from British Columbia and the Northwest Territories were virtually identical to each other and to sequences of *B. vernus* collected in Finland. The sequences were substantially different [much greater than 2% (Zhou et al. 2009; Webb et al. 2012; Cordero et al. 2017)] from other *B. vernus* group members, including group members found in North America (Fig. 1). Morphological examination of the Northwest Territories and Crooked River, BC, specimens revealed traits similar to *B. brunneicolor,* such that the specimens keyed to *B. brunneicolor* in the most recent key to *Baetis* spp. in North America (Wiersema *et al.* 2004, updated with recent couplet patches found in Jacobus *et al.* 2014).

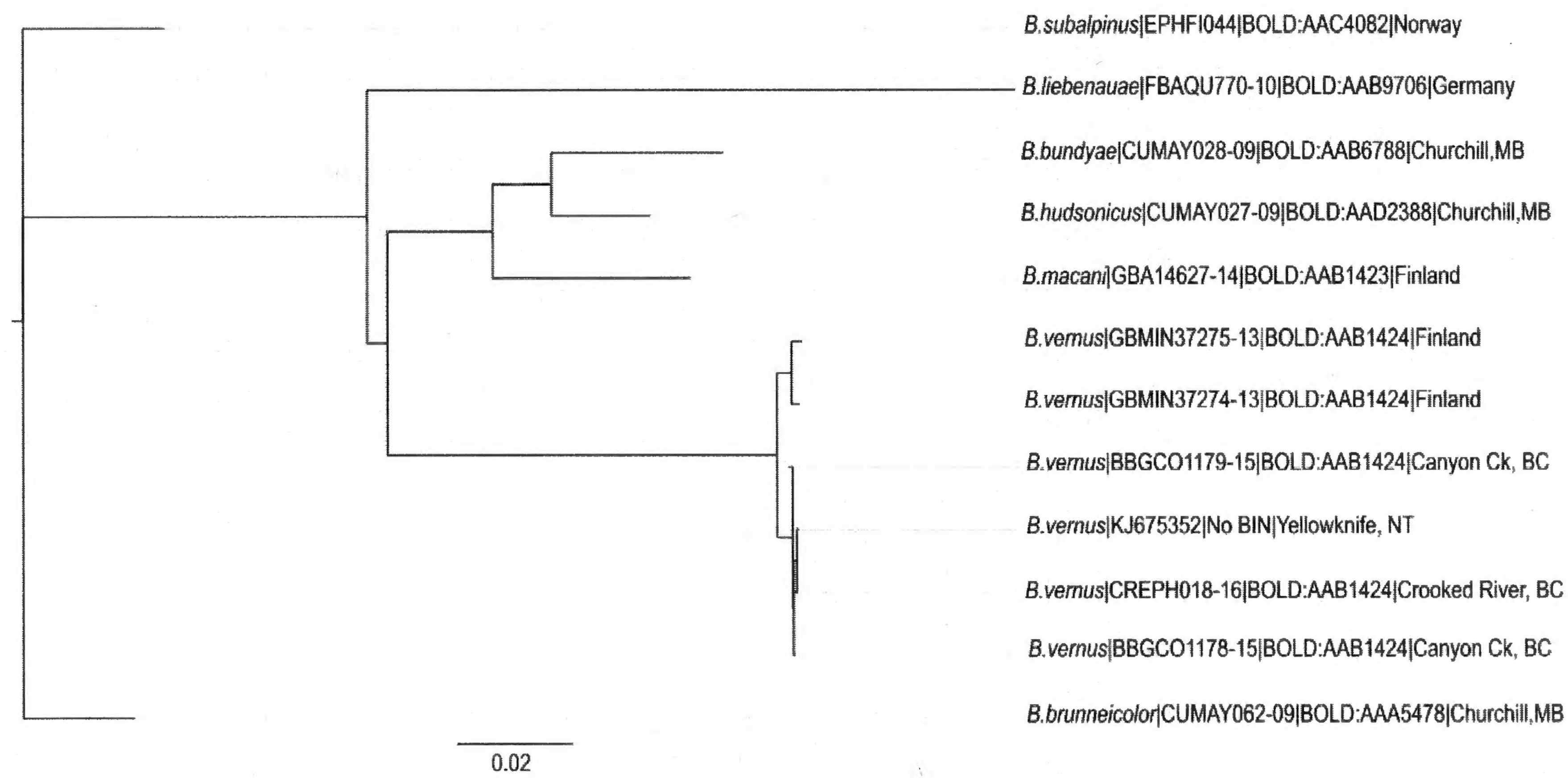

Figure 1: A DNA barcode comparison of Palearctic and Nearctic specimens of *B. vernus* and other closely related baetid species derived from our own collections (Yellowknife and Crooked River *B. vernus* specimens) and from the BOLD database (Ratnasingham and Hebert 2017). The sequences were aligned with Clustal W and visualized with FigTree 1.4.3. Approximate collection locations in Canada and Europe are listed next to each specimen along with that specimen's BIN (Ratnasingham and Hebert 2013). Sequence data are publicly available using the BOLD IDs (most specimens) or NCBI accession number (Yellowknife specimen) associated with each specimen.

These combined results prompted us to look at other *B. vernus* group specimens collected in northwest Canada. These consisted of specimens labeled "*B. brunneicolor*", "*B. bundyae*", and "*Baetis* n. sp. (*vernus* group)" collected from northern Yukon (Porcupine River drainage), southern Yukon (streams along the Alaska Highway), and streams in the Mackenzie Mountains west of the Mackenzie River Valley in the Northwest Territories (Table 1). *Baetis bundyae* and *B. hudsonicus* specimens were usually easy to distinguish from *B. brunneicolor* and *B. vernus* due to the presence of narrow gills on the abdomen. A third group of specimens collected from northern Yukon and from the Mackenzie Mountains west of the Mackenzie River (Table 1) appeared to show characteristics of both groups, with narrow gills like *B. bundyae* but other characters more consistent with *B. vernus.* Due to the age and/or storage conditions of these latter specimens, DNA barcoding was not possible, so the primary concern was distinguishing the larvae of the widespread Nearctic *B. brunneicolor* and the newly discovered *B. vernus*. Morphological features are described for each species below,

including a summary of characters that can be used to distinguish the two species in northwestern Canada.

***Baetis brunneicolor* – General Morphological Description of Larva** (Figs. 2–13):

Head: Frons – General shape subtriangular with blunt apex, lateral edges straight or slightly concave (Fig. 2). **Antennae** – Scape and pedicel with many small hair-like setae, no robust setae present. Small hair-like setae seem to be restricted to distal half of scape (Figs. 3a, 3b). No apparent pattern of small setae on pedicel.

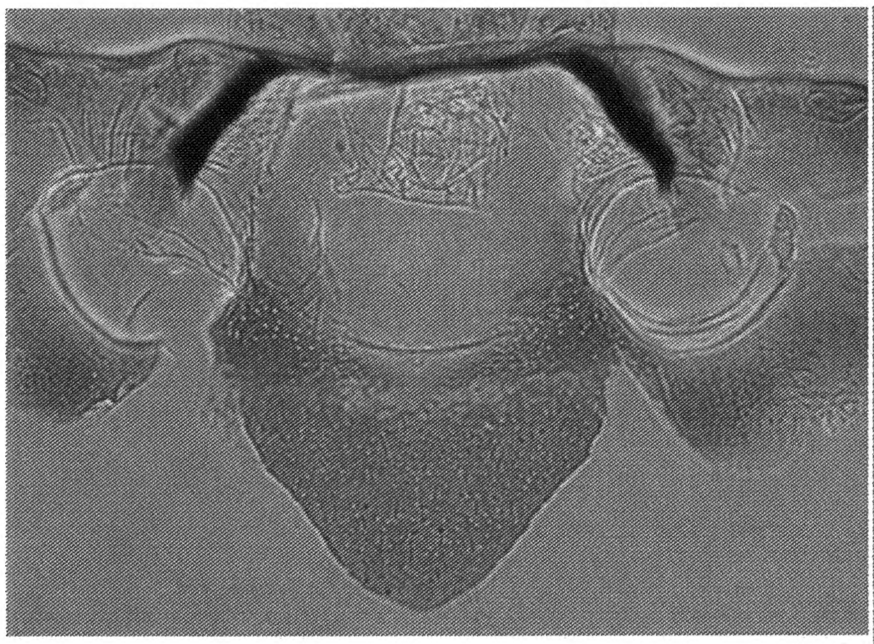

Figure 2. *Baetis brunneicolor*: dorsal view of frons.

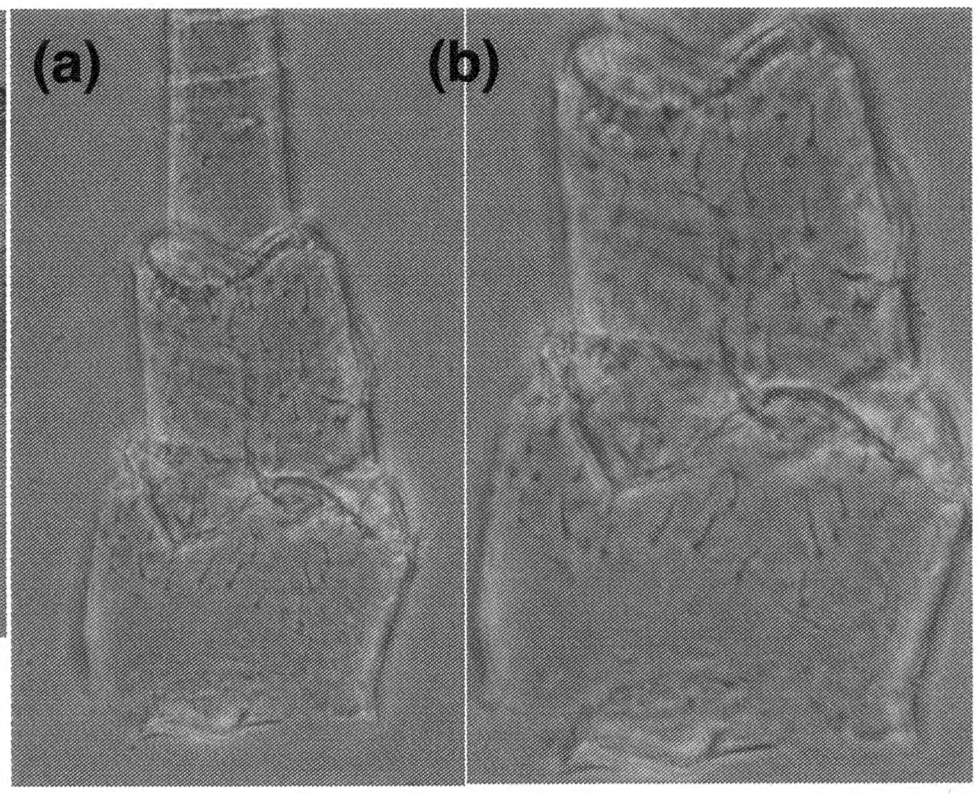

Figure 3. *Baetis brunneicolor*: antennal scape and pedicel; (b) is enlargement of lower section of (a).

Mouthparts: Labrum – Lateral edges tend to be rounded, never appearing straight (Fig. 4a). Dorsal setal pattern 1 long median pair, a gap then row of 4–5 smaller setae (Fig. 4b) extending to edge of anterior margin (i.e., 1 + 4–5). Dorsal surface with many setae, most concentrated near posterior corners. Middle of dorsal surface with somewhat rounded raised area surrounded with small surface setae, no obvious dark marking associated with raised medial area. **Right Mandible** – First tooth of outer incisor larger than second tooth and with squared-off outer edge; second tooth larger than third tooth, and with blunt outer edge; and third tooth smallest of three with rounded outer edge (Fig. 5a, left). This is the "new" condition after moulting, worn teeth are much more similar in size and shape (Fig. 5a, right). Prostheca with pectinate tip, most apical setae about same size and equally spaced (Fig. 5b). **Left Mandible** – One or two small auxiliary teeth present between molar teeth and large apical projection on anterior margin (Fig. 6a). Outer incisor with first tooth slightly larger than second tooth and with squared-off edge; second tooth distinctly larger than third tooth and both teeth with irregularly pointed apices (Fig. 6b, left). This is the "new" condition after moulting, worn teeth are much more similar in size and shape (Fig. 6b, right). **Maxillae** – Four maxillary canines present that lack serrations (Fig. 7a, 7b). Dense brush of long setae along anterior margin of galea-lacinia below canines; margin below setae slightly concave (Fig. 7b). Maxillary palpi two segmented and both segments with many small hair-like setae (Fig. 7a, 7c). Segment 1 of maxillary palpi as long as segment 2. Tips of maxillary palpi extend about one-third of their total length above the tips of canines. **Labium** – Paraglossae broad with mostly straight margins approaching the apices (Fig. 8a, 8b). Apices of paraglossae with 12–15 long setae in two rows. Glossae with broadly pointed apices, ventral surface with single row of about nine long setae located along medial edge (Fig. 8b). Segment 2 of labial palpi with either well developed inner apical lobe and distinctly concaved

margin below lobe (Fig. 8a) or moderately developed inner apical lobe and only slightly concave margin below lobe (Fig. 8c).

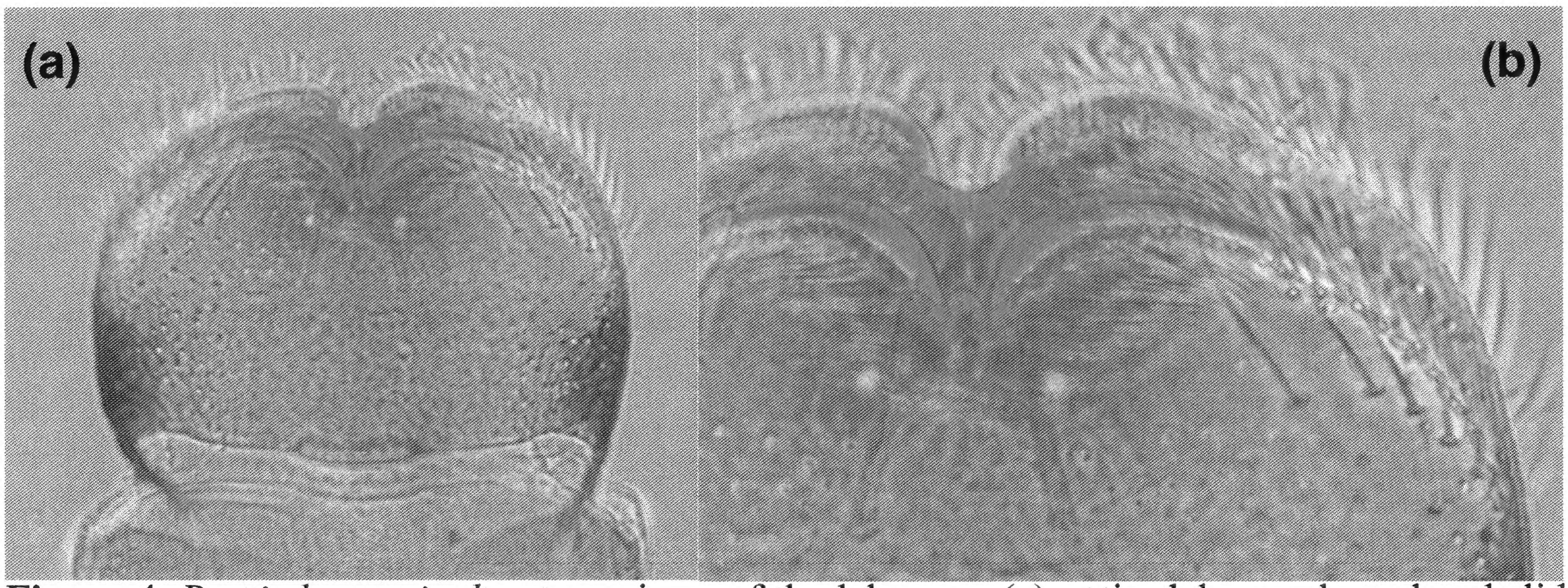

Figure 4. *Baetis brunneicolor*: two views of the labrum – (a) entire labrum cleared and slide-mounted; (b) anterior area enlarged to show setal pattern.

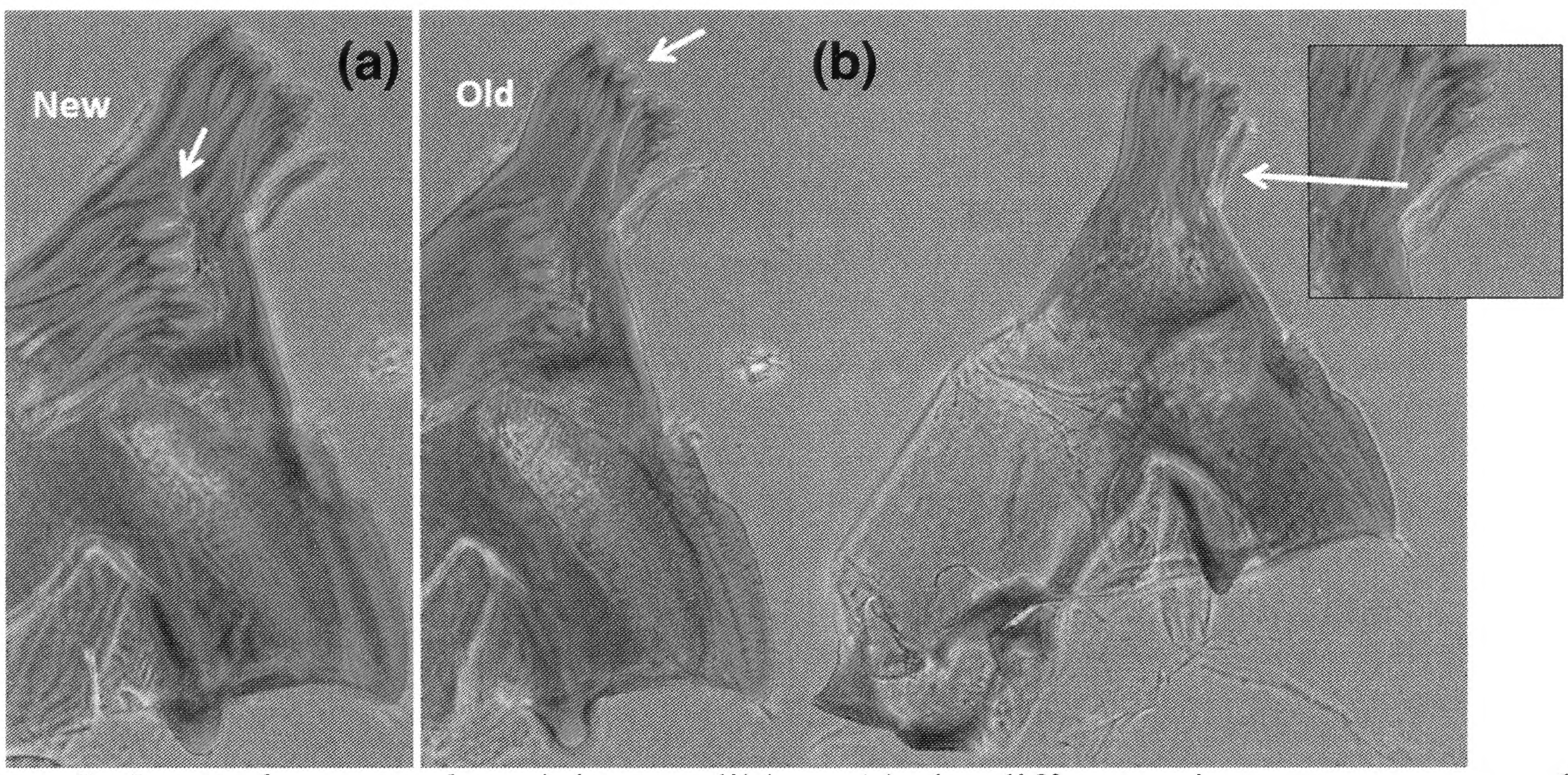

Figure 5: *Baetis brunneicolor*: right mandible – (a) the difference in wear on new and old incisors (new incisors of next instar visible in cleared mandible); (b) entire mandible with prostheca enlarged in inset.

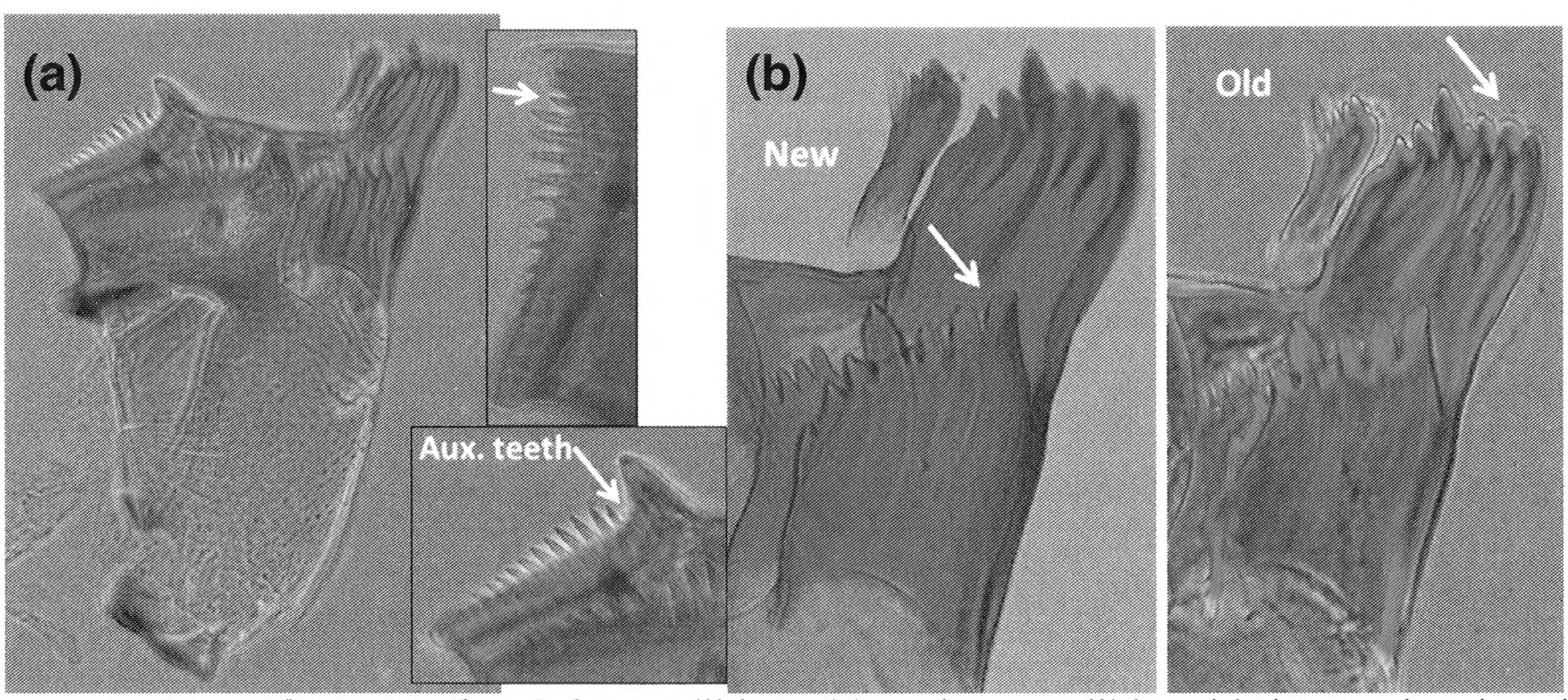

Figure 6. *Baetis brunneicolor*: left mandible – (a) entire mandible with insets showing the auxiliary teeth; (b) the difference in wear on new and old incisors (new incisors of next instar visible in cleared mandible).

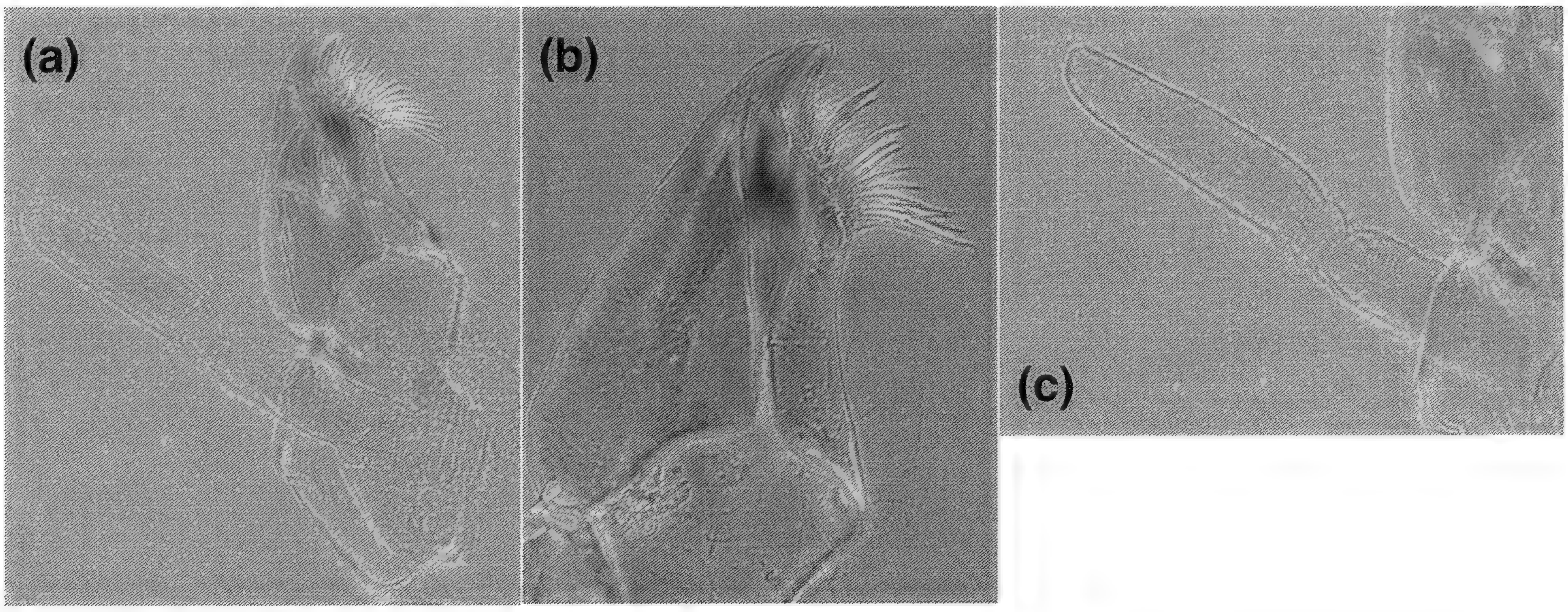

Figure 7. *Baetis brunneicolor*: maxilla – (a) entire maxilla; (b) canines; (c) maxillary palp.

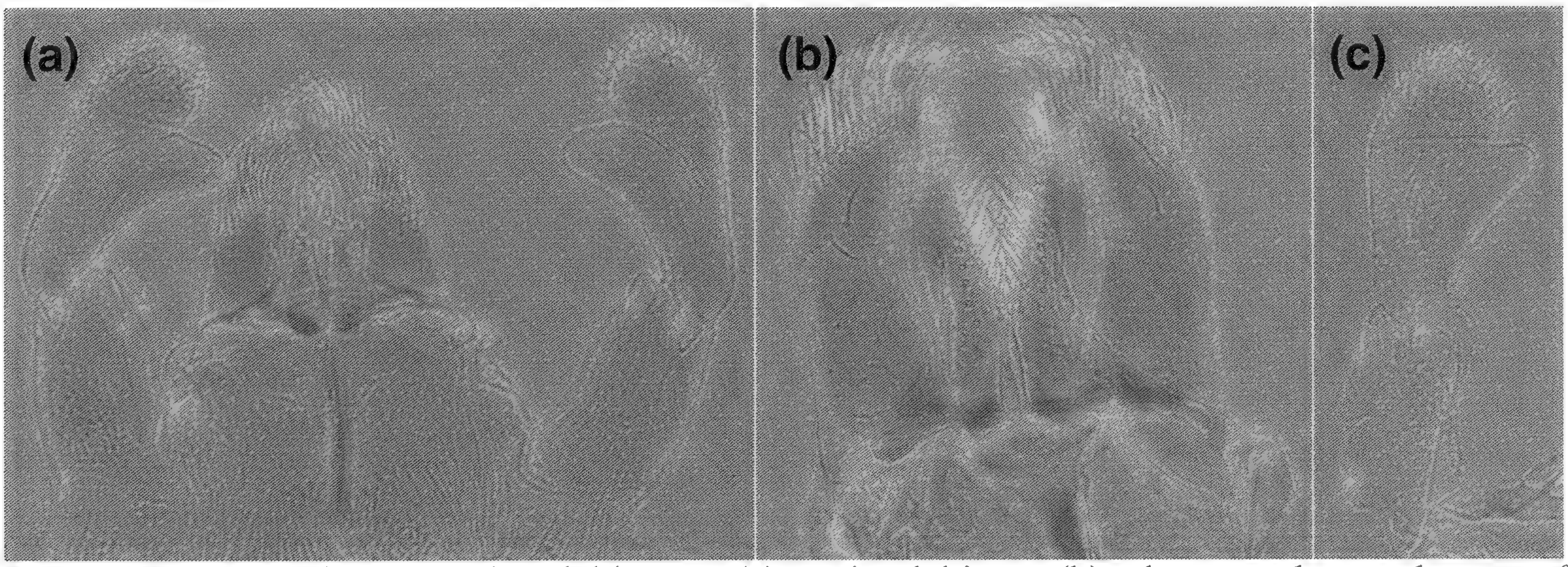

Figure 8. *Baetis brunneicolor*: labium – (a) entire labium; (b) glossa and paraglossae of labium; (c) labial palp.

Forelegs: Femora broadest near midpoint of segment (Fig. 9a). Outer edge with two staggered rows of long, blunt setae that have uniform width from base to tip, setae become more widely spaced and fewer in number near apex of segment (Fig. 9a). Tibia and tarsus generally seem to be much stouter (i.e., wider and shorter) compared to those of *B. vernus* (compare Fig. 9b to Fig. 21b). Foreleg claw with about nine denticles that progressively become larger from base toward apex, apex of claw appears slightly attenuated (i.e., narrowed) (Fig. 9b).

Abdomen: Abdominal Tergite V – Shape typical for abdomen with outer edge of tergite slightly tapering posteriorly (Fig. 10, top). Posterior lateral corners with minimal dark brown colour in fresh specimens at gill insertion. Numerous scale setae present and few scattered hair-like setae between scale setae. Surface with faint cuticular ridges (i.e., weakly grainy) (Fig. 10, bottom). Posterior margins with spinules, but not darkly pigmented compared to rest of tergite (Fig. 10, top). **Abdominal Gills 4 and 5** – Gill 4 larger than gill 5, but both have same basic oval shape with smoothly curved dorsal edge and outer margin (Fig. 11). Both gills have marginal teeth; at 20X magnification gill 4 has about 8 teeth/0.05 mm of edge and gill 5 has 8–9 teeth/0.05 mm of edge. Both gills have distinct central trachea with one or two smaller side branches visible (Figs. 11). Gill 4 length slightly less than twice the width (i.e., width x 1.8=length). Gill 5 has same length/width relationship as gill 4. **Paraprocts** – Inner apical edges with regular, large spines (Fig. 12). Surface with scattered long, hair-like setae that are more or less uniformly distributed over surface (Fig. 12). No other distinctive surface features or textures.

Colour Pattern of Body: Overall body colour uniform brown with less distinct contrasting lighter areas (Fig. 13a). Pronotum lacking bi-lobed brown spots, but large somewhat "c-shaped" diffuse blotches are sometimes present (Fig. 13b). Meso- and metanotum of thorax mostly brown with some lighter streaks and spots, especially on the mesonotum. Abdominal terga with submedian paired brown spots; these are faint on some specimens (Fig. 13a). A medial pale spot with paired lateral pale spots separated by brown background colour seems a common pattern on terga. Posterolateral edges of terga pale compared to brown medial part of terga.

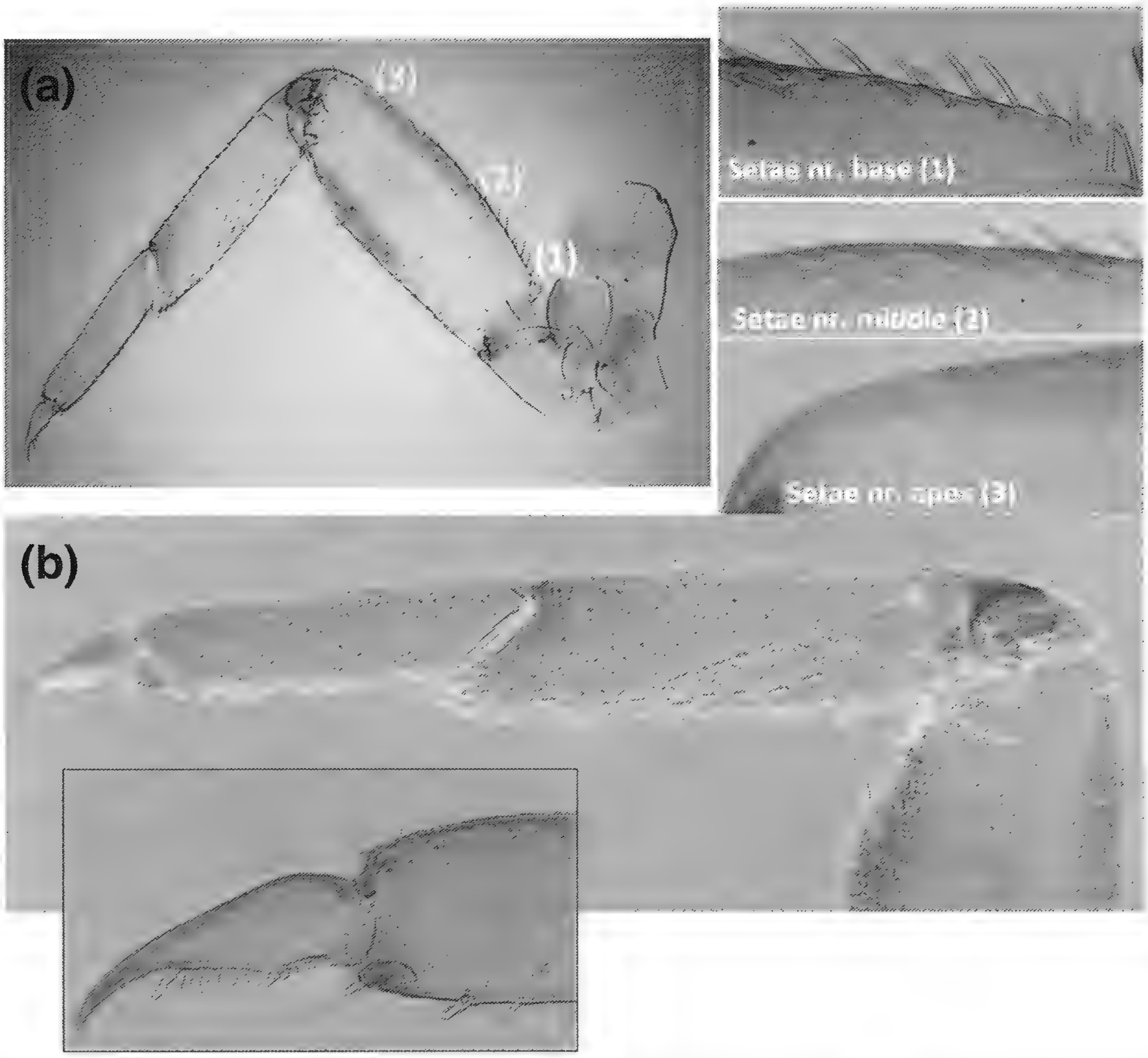

Figure 9. *Baetis brunneicolor*: foreleg – (a) entire foreleg, with numbers denoting areas of the femur with setal patterns shown at right; (b) tibia and tarsus, with inset showing denticles on claw.

General Shape of Abdomen: The overall shape of the abdomen, viewed dorsally, is one of a gradually tapering cylinder that is widest at segment I and narrowest at segment X (Fig. 13a). The shape results from a change in the width/length ration from anterior segments to posterior segments. Segment I is about three times as wide as long and segment X is almost as wide as long.

***Baetis vernus* – General Morphological Description of Larva** (Figs. 14–26):

Head: Frons – General shape subtriangular with blunt apex, lateral edges either straight or slightly concave (without slide mounting intact specimens can even appear slightly convex) (Fig. 14). **Antennae** – Scape and pedicel with many small hair-like setae, no robust setae present. Small hair-like setae seem to be restricted to distal part of scape (Fig. 15). No apparent pattern of small setae on pedicel.

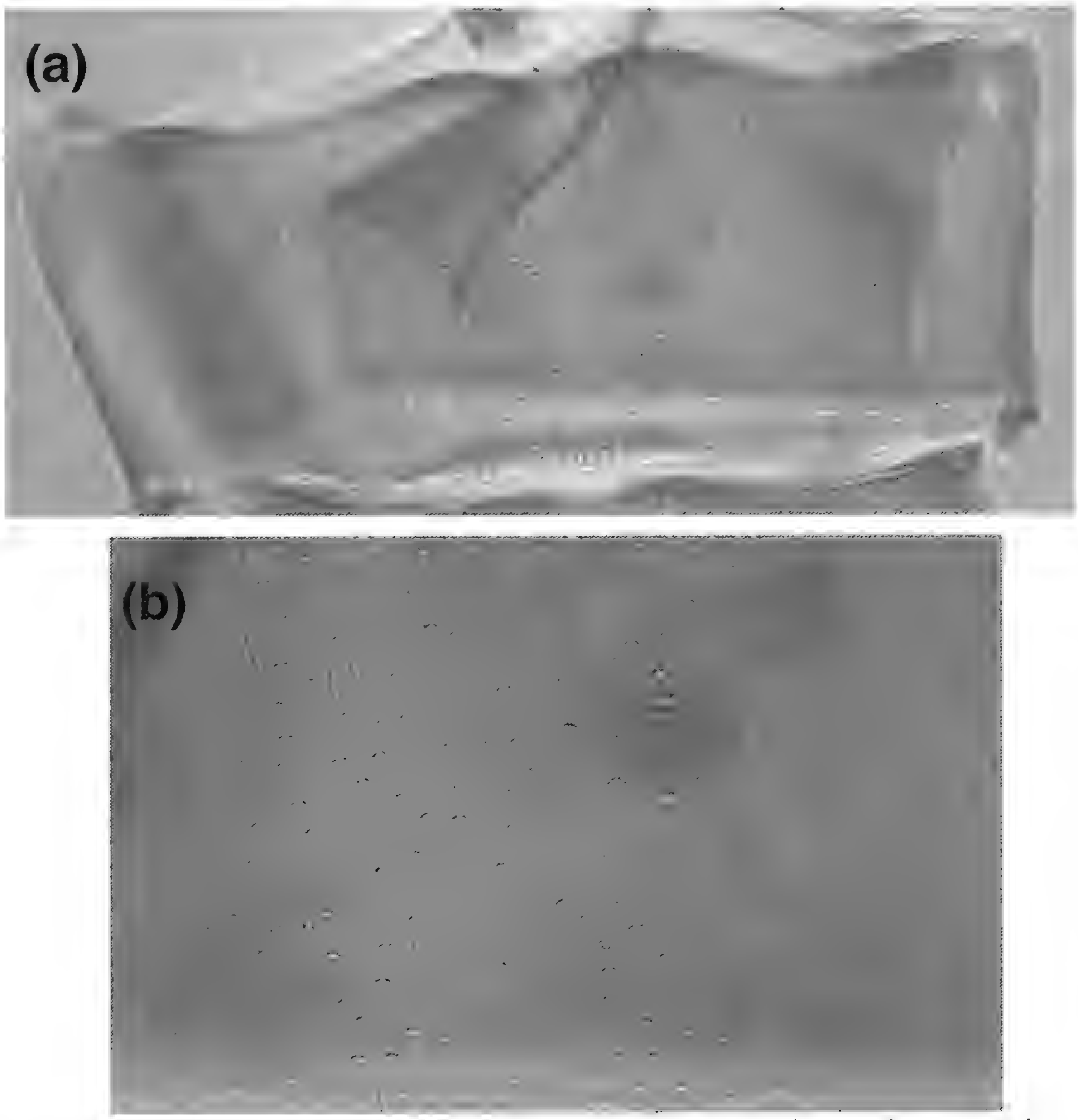

Figure 10. *Baetis brunneicolor*: Abdominal tergite V – (a) entire tergite; (b) enlargement showing cuticular patterns.

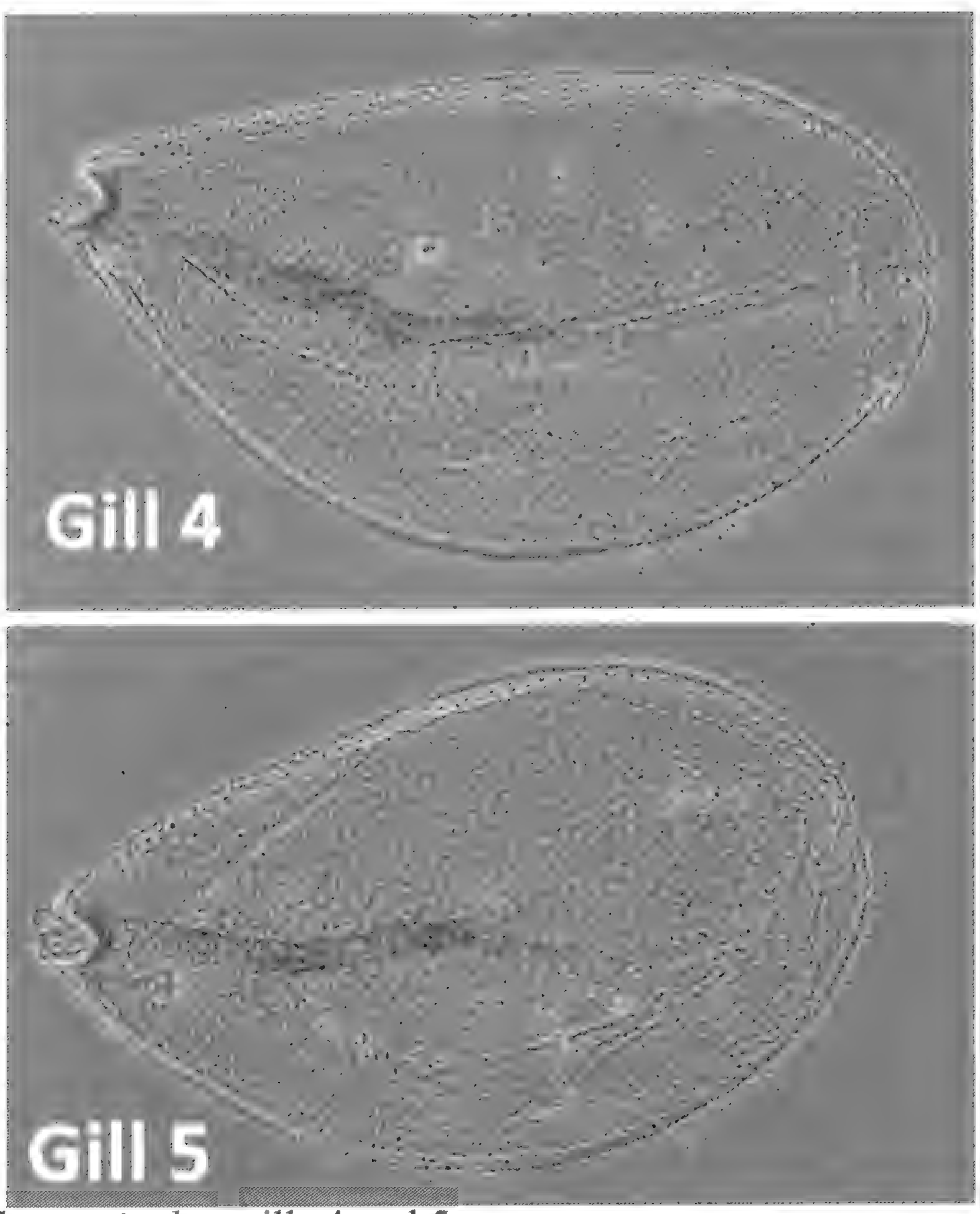

Figure 11. *Baetis brunneicolor*: gills 4 and 5.

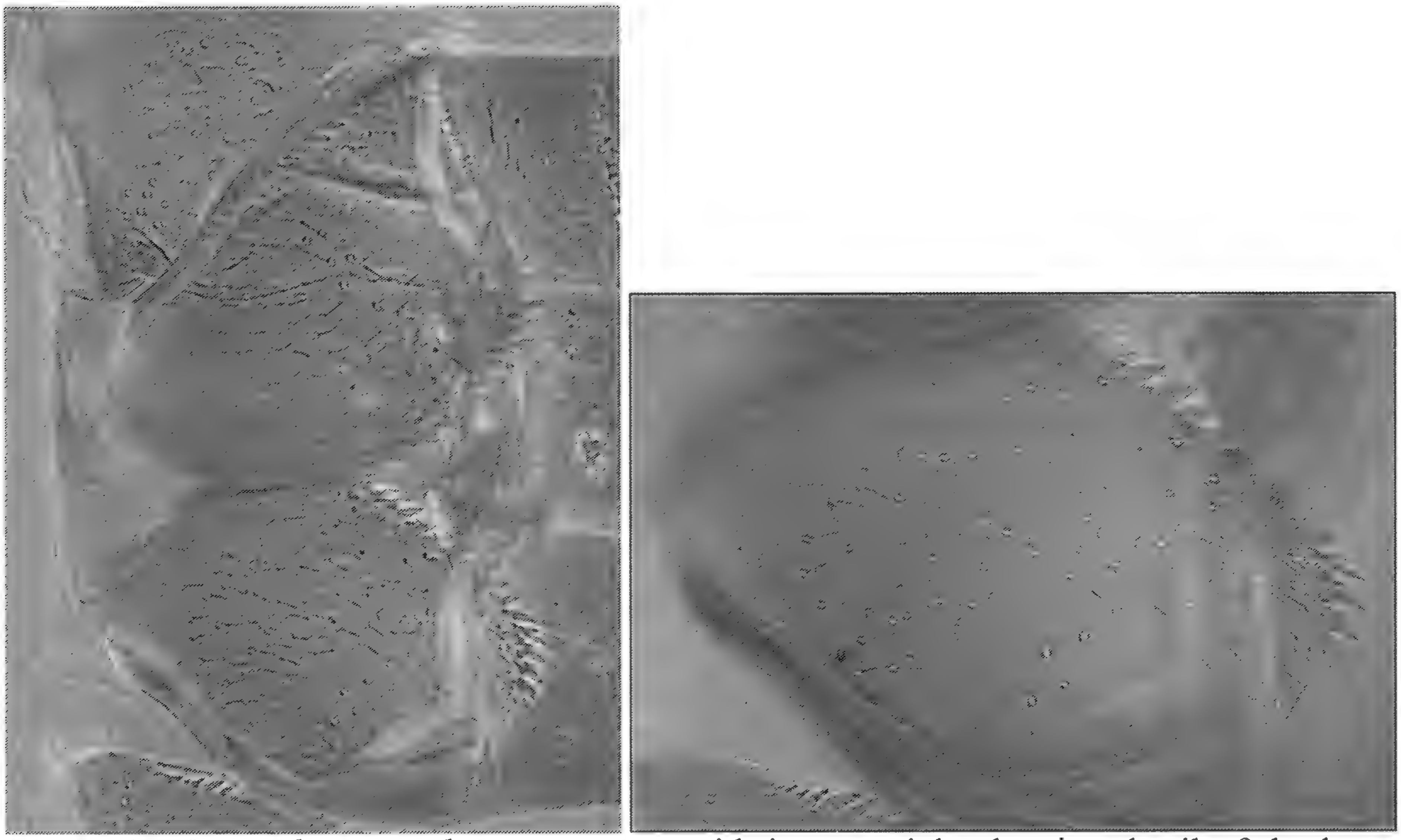

Figure 12. *Baetis brunneicolor*: paraprocts, with inset at right showing detail of the lower paraproct at left.

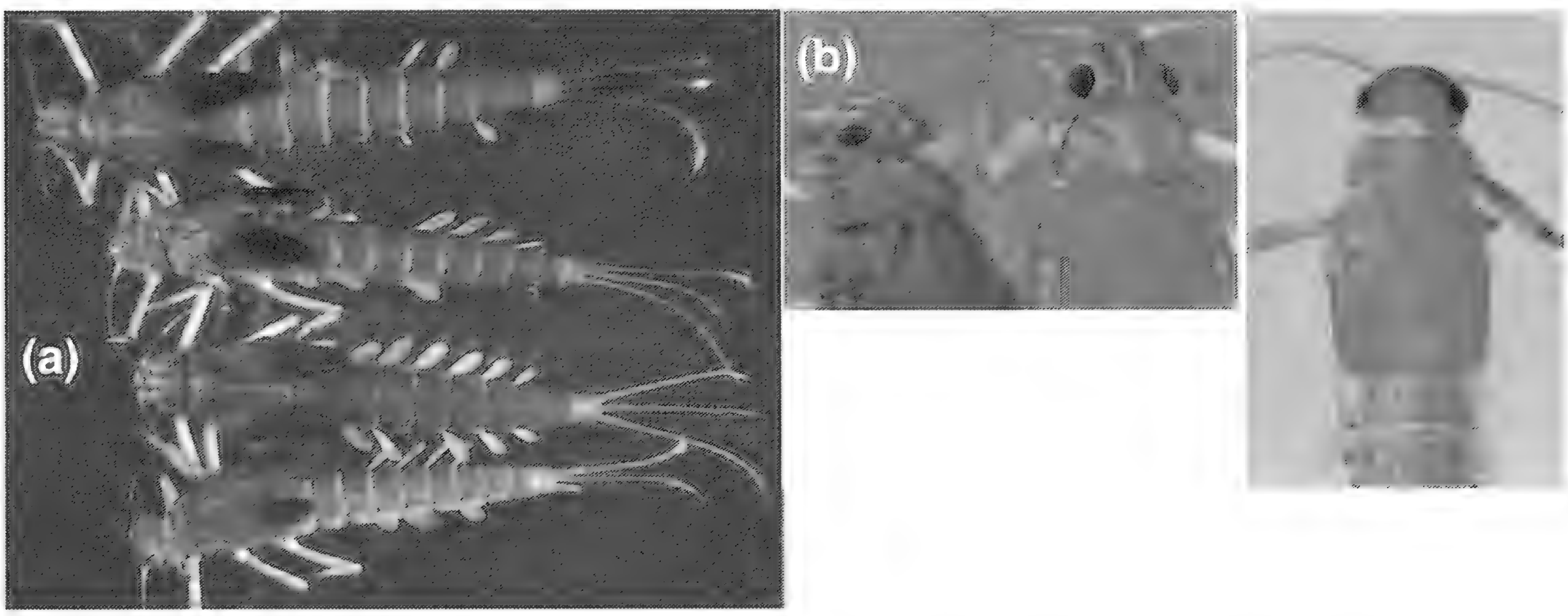

Figure 13. *Baetis brunneicolor*: body features – (a) dorsal views of several larvae, showing colour patterns and body shape; (b) detail of anterior sections of larvae showing colour patterns on thorax.

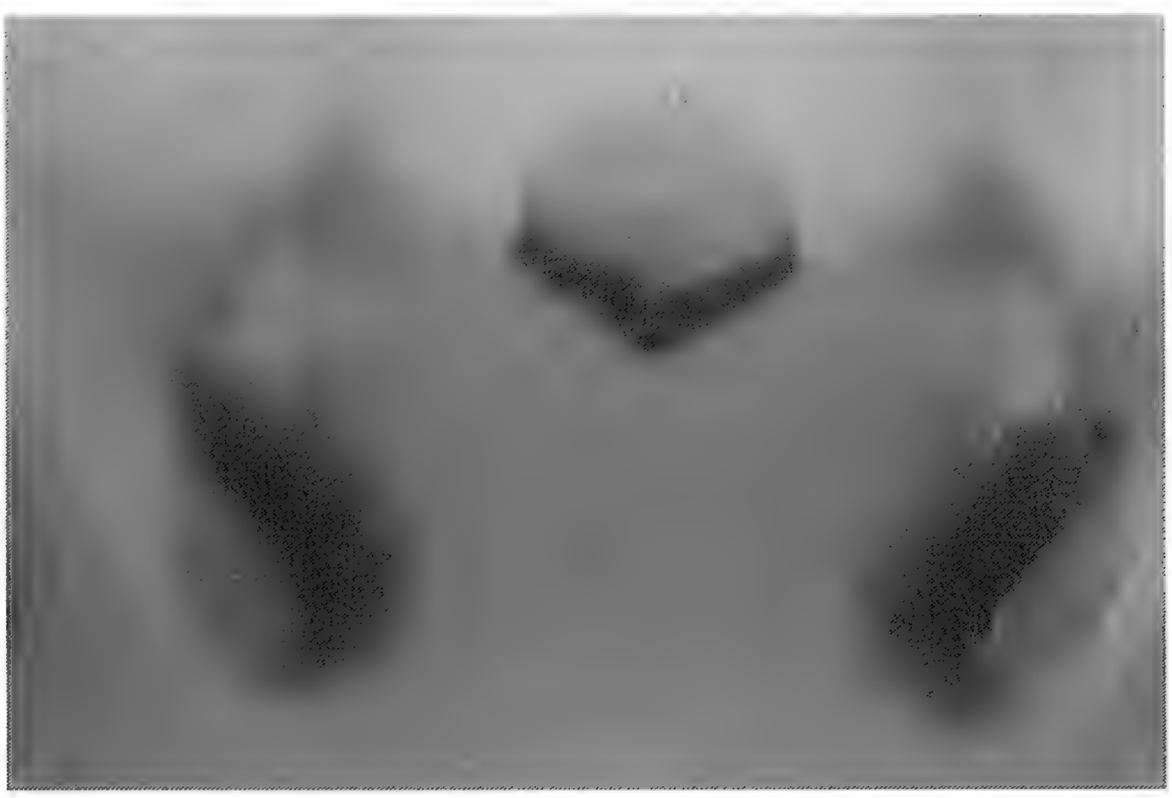

Figure 14. *Baetis vernus*: dorsal view of frons.

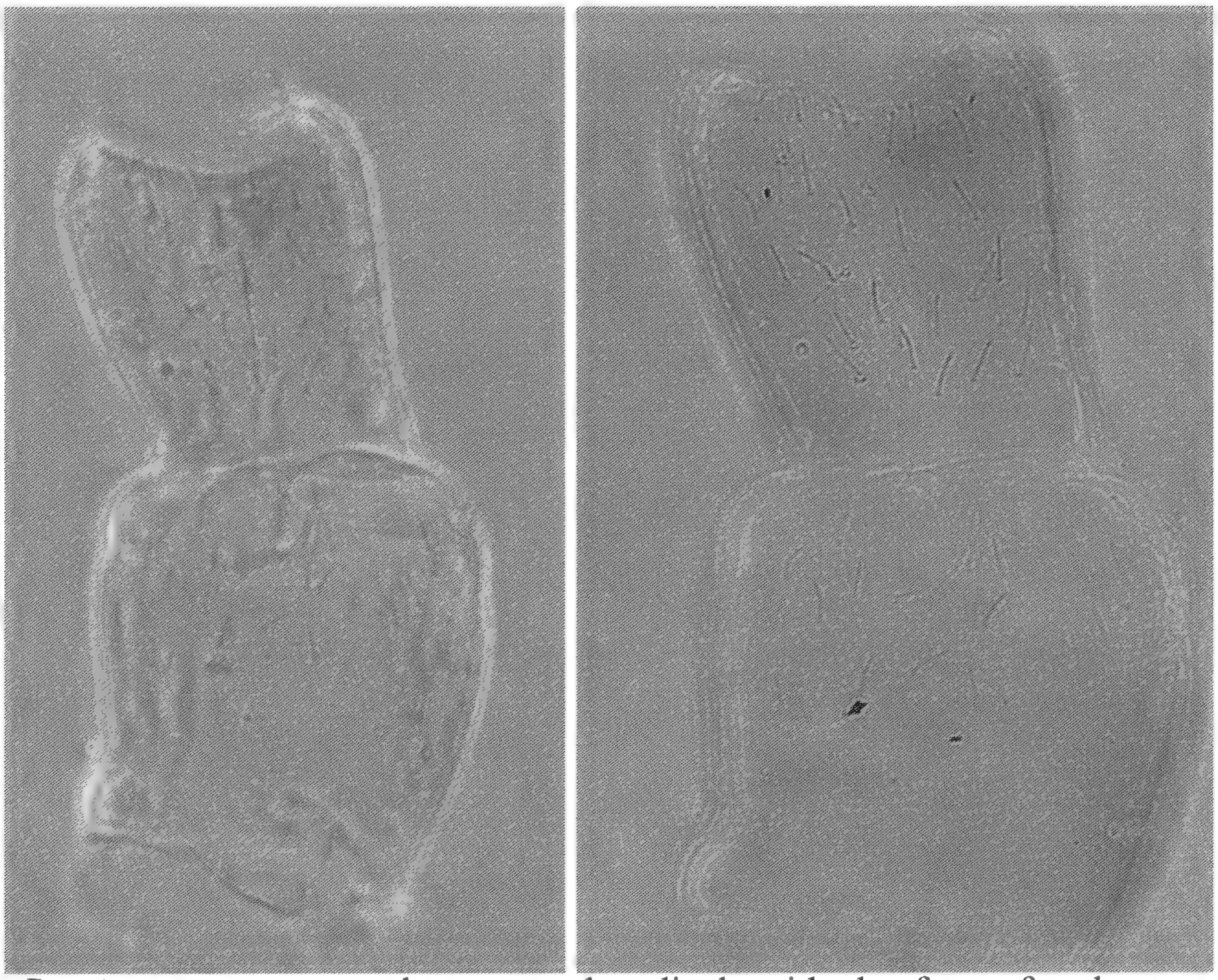

Figure 15. *Baetis vernus*: antennal scape and pedicel, with the focus for the two images highlighting different features.

Mouthparts: Labrum – Lateral edges tend to be straight, or at most slightly rounded (Fig. 16a), making the labrum appear somewhat rectangular. Dorsal setal pattern 1 long medial pair, a gap then row of 3–4 smaller setae (Fig. 16b) extending to edge of anterior margin (i.e., 1 + 3–4). Dorsal surface with relatively few scattered setae, most tend to be concentrated near edges of somewhat triangular raised area that is flanked by dark bands (bands faint on some specimens) (Fig. 16a). **Right Mandible** – First tooth of outer incisor larger than second tooth and with squared-off outer edge; second tooth only slightly larger than third tooth and both with irregularly pointed tips (Fig.17a, left). This is the "new" condition after moulting, worn teeth are much more similar in size and shape (Fig. 17a, right). Prostheca pectinate with a single row of setae along inner edge near apex (Fig. 17b). Setae of variable lengths and some form a cluster near apex of prostheca. **Left Mandible** – One or two small auxiliary teeth present between molar teeth and large apical projection on anterior margin (Fig. 18a). Outer incisor with first tooth slightly larger than second tooth and with squared-off edge; second tooth only slightly larger than third tooth and both teeth with irregularly pointed apices (Fig. 18b, left). This is the "new" condition after moulting, worn teeth are much more similar in size and shape (Fig. 18b, right). **Maxillae** – Four maxillary canines present that lack serrations (Figs. 19a, 19b). A dense brush of long setae along anterior margin of galea-lacinia below canines; margin below setae straight or only slightly concave (Figs. 19a, 19b). Maxillary palpi two segmented and both segments with many small hair-like setae (Fig. 19a). Segment 1 of maxillary palpi as long as segment 2. Tips of maxillary palpi extend about one-third of their total length above the tips of canines (Fig. 19a). **Labium** – Paraglossae with broad curved apices (Figs. 20a, 20b). Apices of paraglossae with 11–12 long setae in two rows (Fig. 20b). Glossae with narrowly pointed apices, ventral surface with single row of about six long setae located along medial edge (Fig. 20b). Segment 2 of labial palpi with moderately developed inner apical lobe and slightly concaved margin below lobe (Fig. 20a).

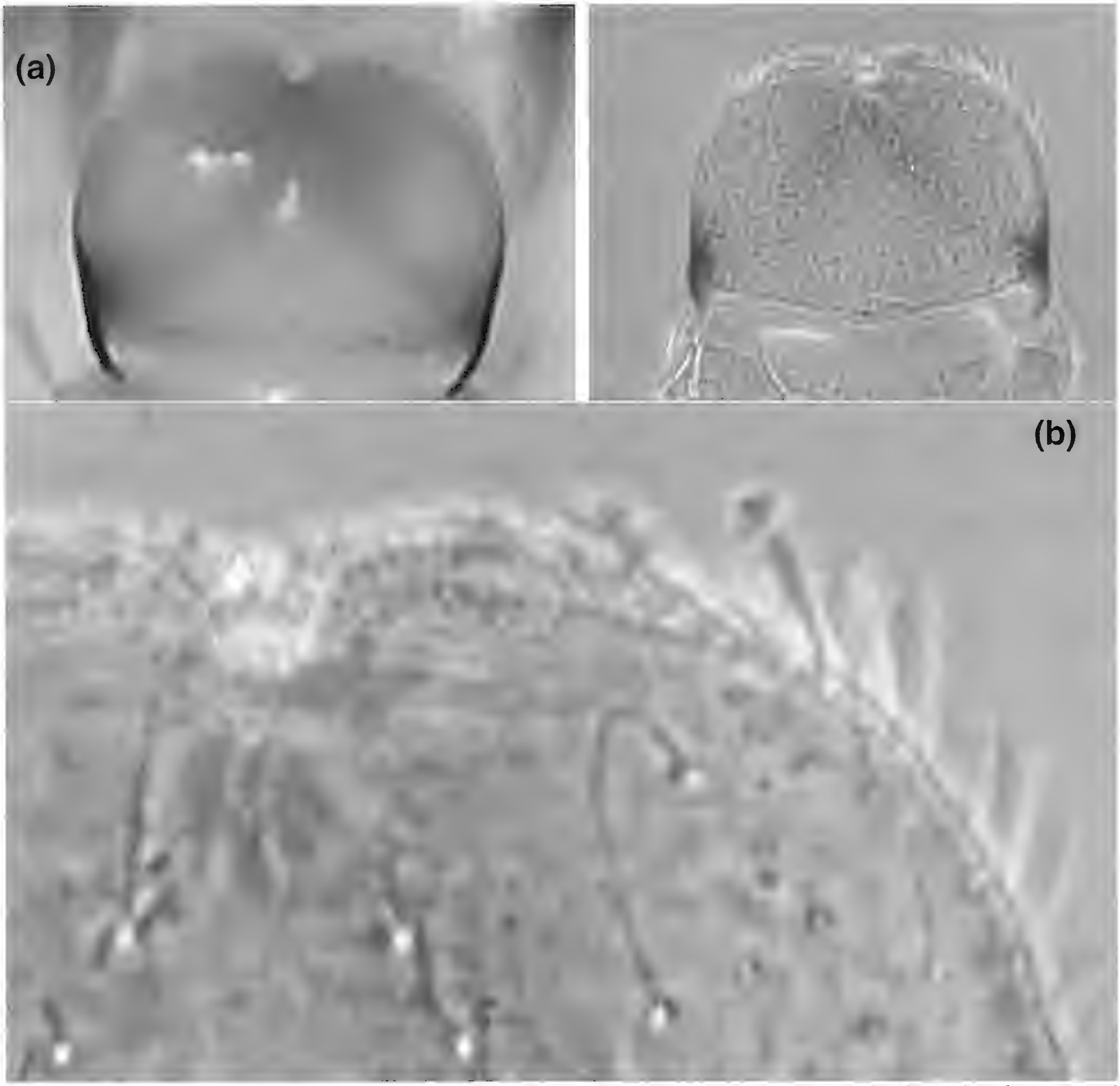

Figure 16. *Baetis vernus*: labrum – (a) left image is the labrum as attached to the frons, and right image is cleared and slide mounted; (b) enlarged portion of labrum showing setal pattern.

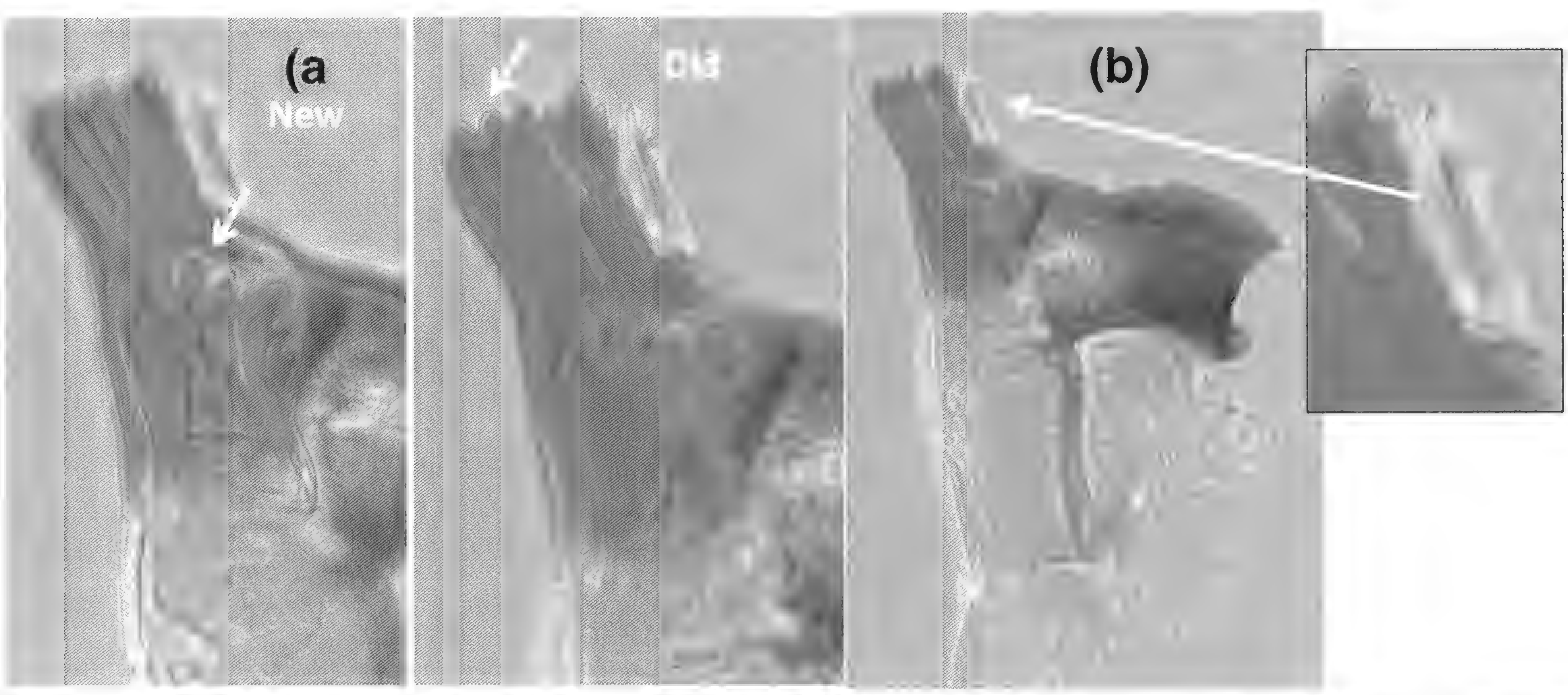

Figure 17. Baetis vernus: right mandible – (a) shows the difference in wear on new and old incisors (new incisors of next instar visible in cleared mandible); (b) entire mandible with inset showing prostheca.

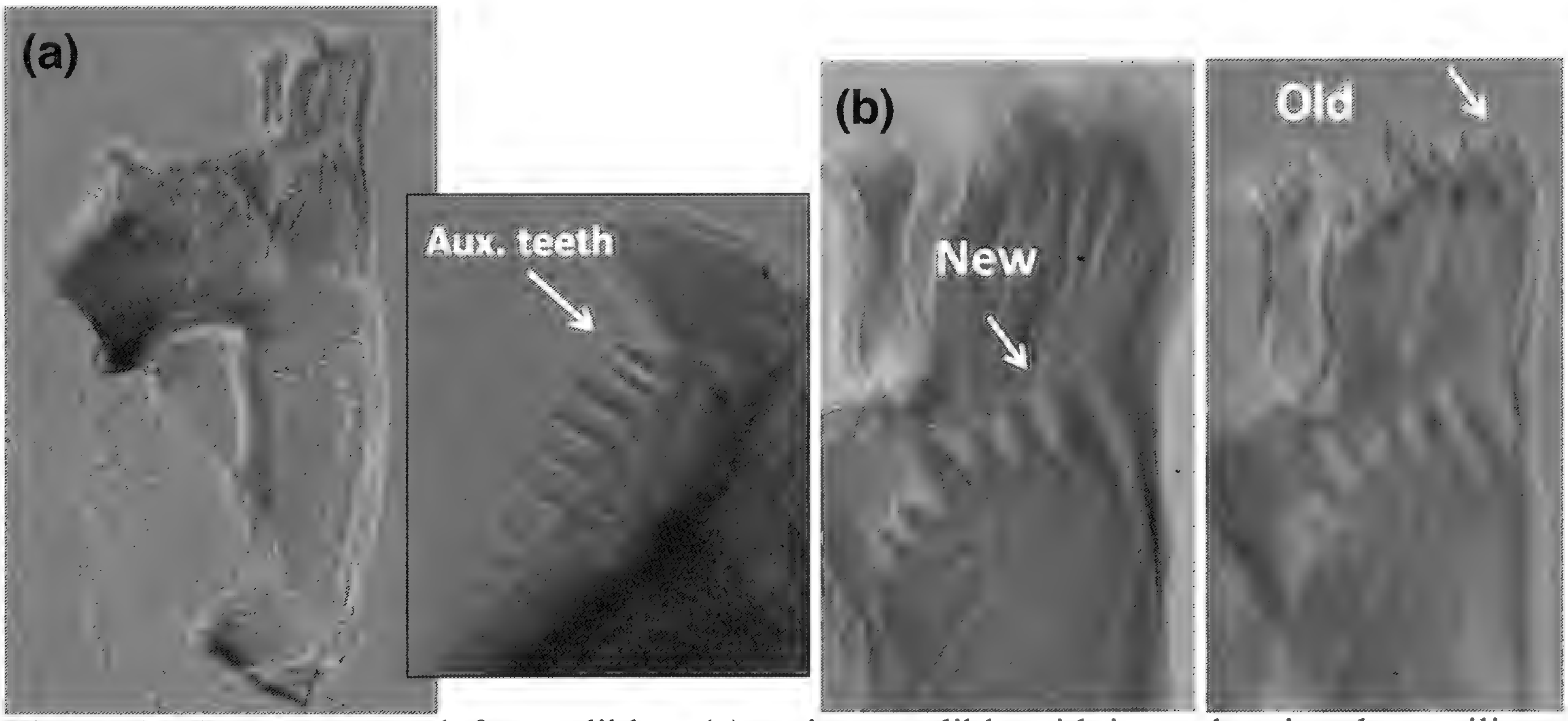

Figure 18. *Baetis vernus*: left mandible – (a) entire mandible with inset showing the auxiliary teeth; (b) the difference in wear on new and old incisors (new incisors of next instar visible in cleared mandible).

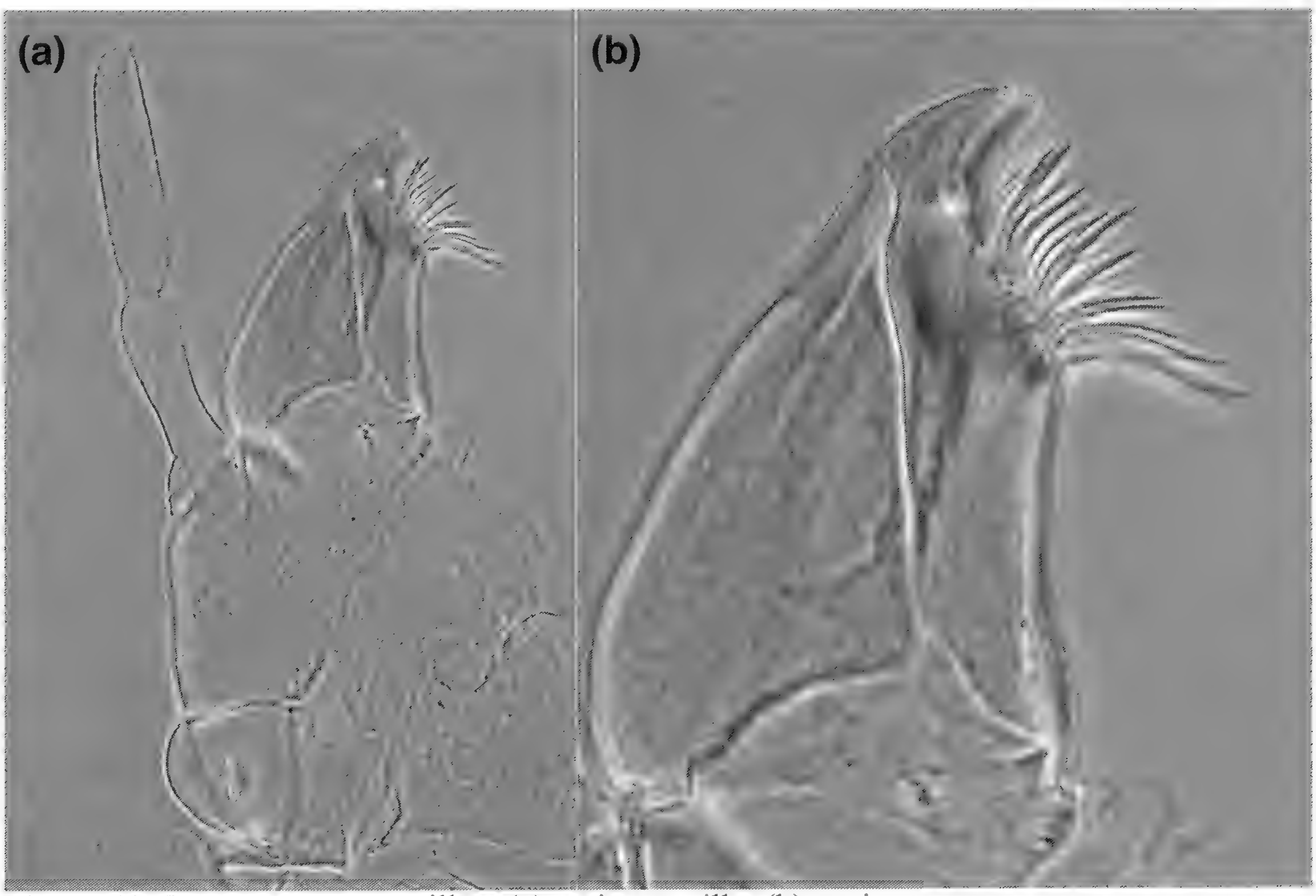

Figure 19. *Baetis vernus*: maxilla – (a) entire maxilla; (b) canines.

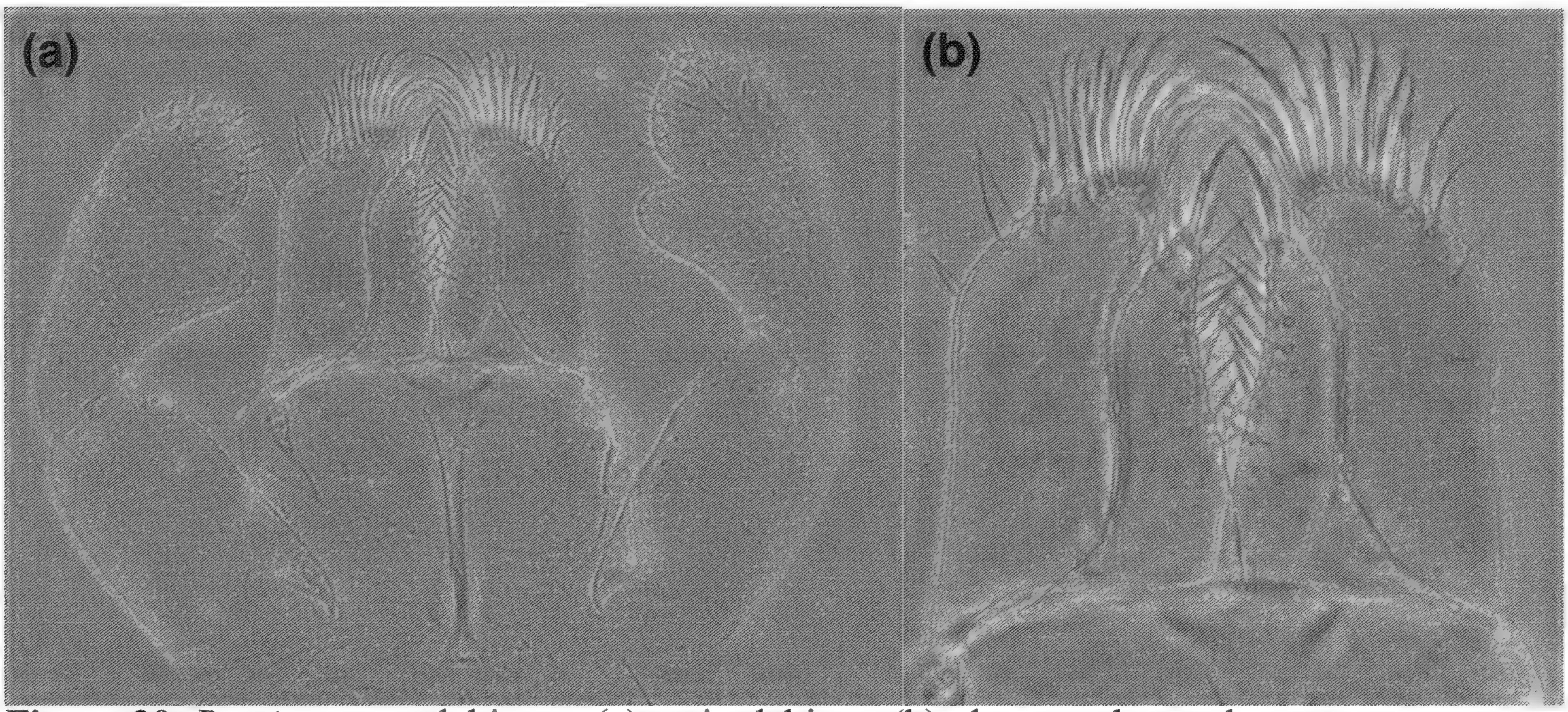

Figure 20. *Baetis vernus*: labium – (a) entire labium; (b) glossa and paraglossa.

Forelegs: Femora about same width from base to apex (Fig. 21a). Outer edge with two staggered rows of long, blunt setae, many of which have narrow bases and broad ends, but some setae have uniform width from base to tip (Fig. 21a). Setae on outer edge of femora are numerous near the base of the segment and gradually become fewer in number approaching joint with tibia, stopping entirely just before the joint with the tibia (Fig. 21a, right hand panels). Tibia and tarsus are thinner and more delicate compared to those of *B. brunneicolor* (compare Fig. 21b to Fig. 9b). Foreclaw with 8–10 denticles, small near base of claw and only gradually become larger toward apex making the row appear more uniform over its length (Fig. 21b). Apex of claw thicker and not noticeably attenuated as in *B. brunneicolor* (compare Figs. 9b and 21b)

Abdomen: Abdominal Tergite V – Shape typical for abdomen with outer edges of tergite nearly parallel (Fig. 22). Posterior lateral corners with distinct dark brown colour at gill insertions (Fig. 22). Numerous scale setae present, but widely spaced over surface of cuticle and with few scattered hair-like setae between scale setae (Fig. 22). Surface with distinct cuticular ridges (i.e., moderately grainy). Posterior margins with spinules, pigmented darker brown compared to lighter brown colour of rest of tergite (Fig. 22). **Abdominal Gills 4 and 5** – Gill 4 is larger than gill 5 and shaped distinctly different shape compared to gill 5 (Fig. 23). Gill 4 more subtriangular with a bulged dorsal margin. Gill 5 closer to sub-oval shape typical of *B. brunneicolor* gills (Fig. 23). Both gills have marginal teeth; at 20X magnification, gill 4 has about 8 teeth/0.05 mm of edge and gill 5 has 8–9 teeth/0.05 mm of edge. Gill 4 has only faint traces of the central trachea and gill 5 has no visible trachea (Fig. 23). Gill 4 almost exactly twice as long as wide. Gill 5 length is slightly less than twice the width. **Paraprocts** – Inner apical edges with irregular row of large spines, which become smaller around the apical corner (Fig. 24). Surface with few scattered hair-like setae and dense cluster of small cuticular scales near outer apical edge (Fig. 24).

Colour Pattern of Body: Overall body colour of Northwest Territories (NT) specimen is much more contrasting compared to the BC specimen (Figs. 25, 26). Generally, body somewhat brown with large pale areas. Pronotum with distinct paired medial brown spots or blotches, lateral edges dark brown, but rest of surface pale (Figs. 25, 26). Thorax with several large pale areas and smaller distinct brown spots or blotches (BC specimen seems closer in thoracic colour patterning to *B. brunneicolor* than NT specimen) (compare Figs. 25, 26 to Fig. 13). Abdominal terga I–IV of NT specimen mostly brown with large paired pale spots and a smaller medial spot (Fig. 25). Tergite V mostly white with limited brown marks at anterior margin and laterally. Terga VI–IX similar in colour to preceding terga. Tergite X white. The BC specimen had a much less-contrasting overall colour pattern but almost the same pattern of marks and spots (Fig.

26). However, tergite VI on the BC specimen was not pale, but patterned similar to other terga. Also, tergite X was uniformly light brown, not pale as in the NT specimen.

General Shape of Abdomen: The overall shape of the abdomen, viewed dorsally, seemed to change more gradually over its length, not appearing distinctly tapered as in *B. brunneicolor* (compare Figs. 25, 26 to Fig. 13). The change in width/length relationship was less per segment, which resulted in the appearance of a more uniformly shaped abdomen. Edges of individual tergites seemed less tapered compared to *B. brunneicolor*. On the BC specimen (Fig. 26), where gills were lost, it was clear that the abdomen did taper from anterior to posterior, segment I was approximately 2.3 times as wide as long. Segment X was slightly wider than long.

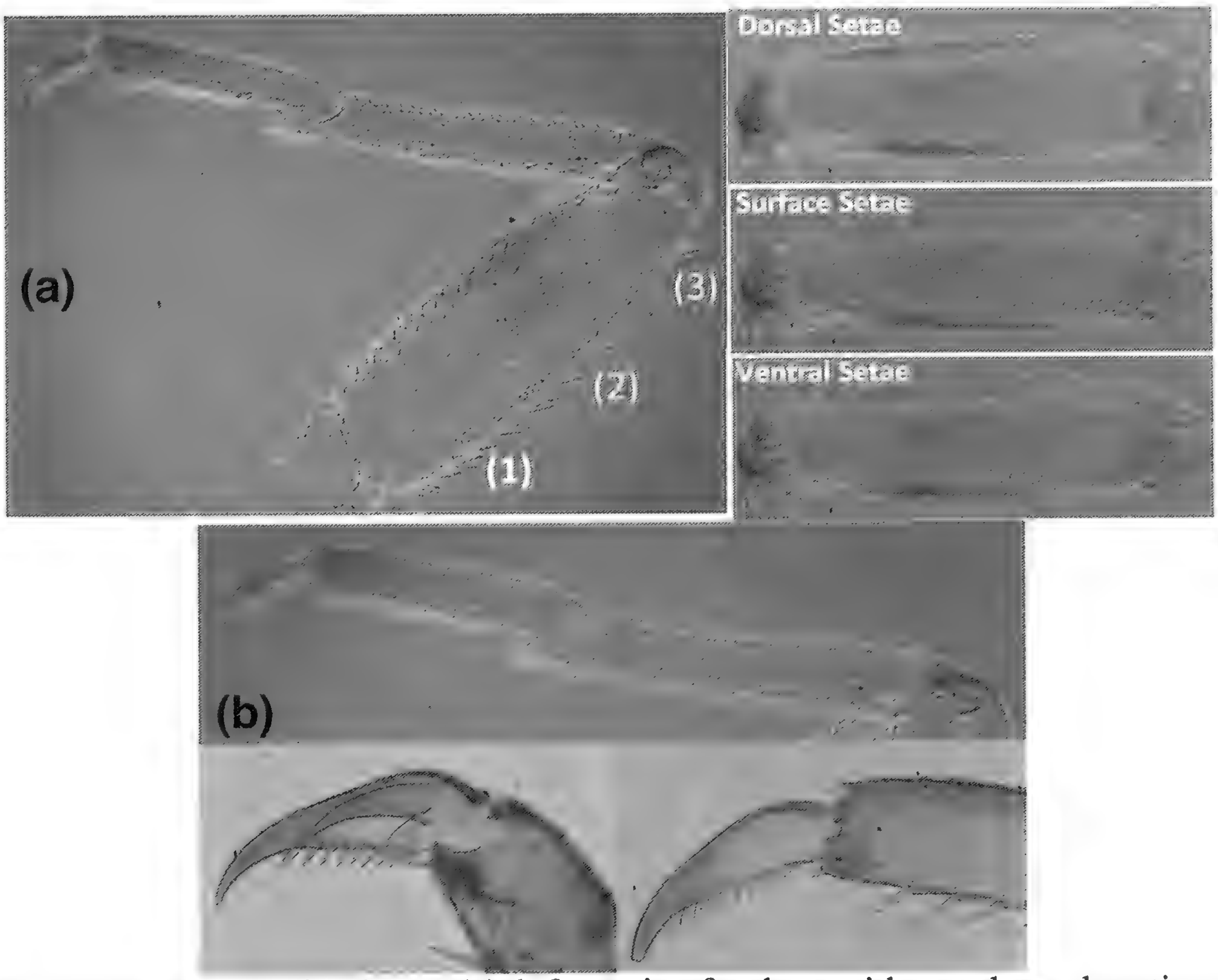

Figure 21. *Baetis vernus*: foreleg – (a) left: entire foreleg, with numbers denoting areas for femur setal patterns; panels at right show the same view at different focus settings to show setal patterns; (b) tibia and tarsus, with insets showing denticles on claw.

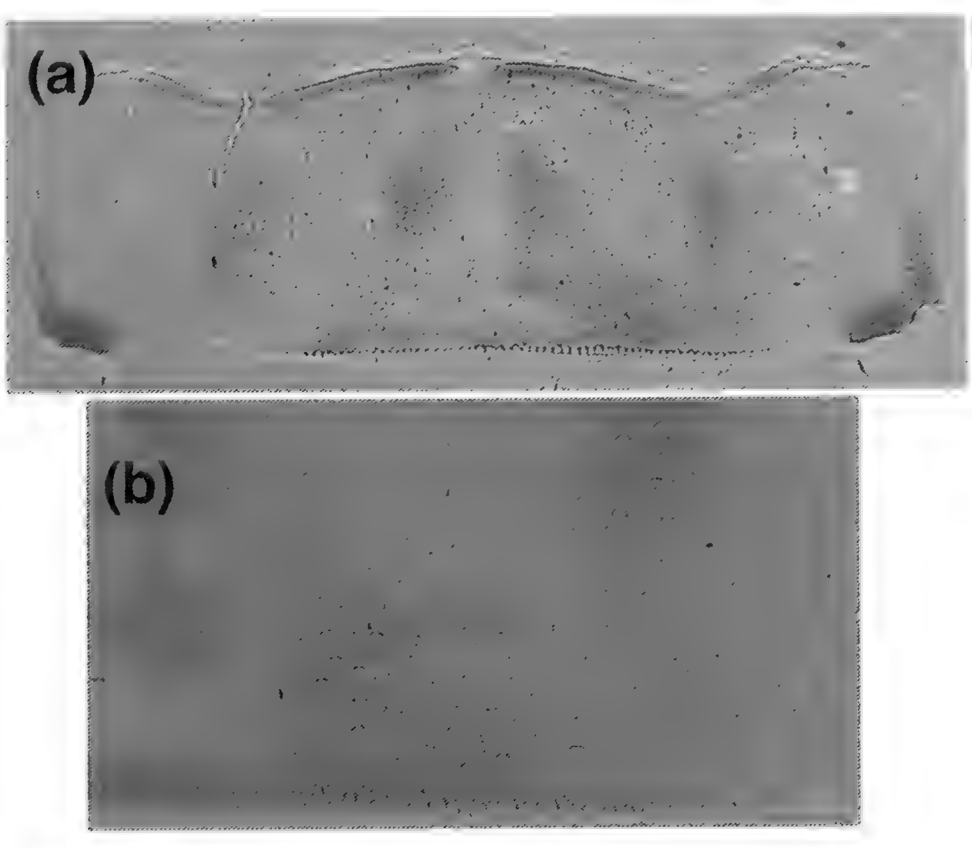

Figure 22. *Baetis vernus*: Abdominal tergite V – (a) entire tergite; (b) enlargement showing cuticular patterns.

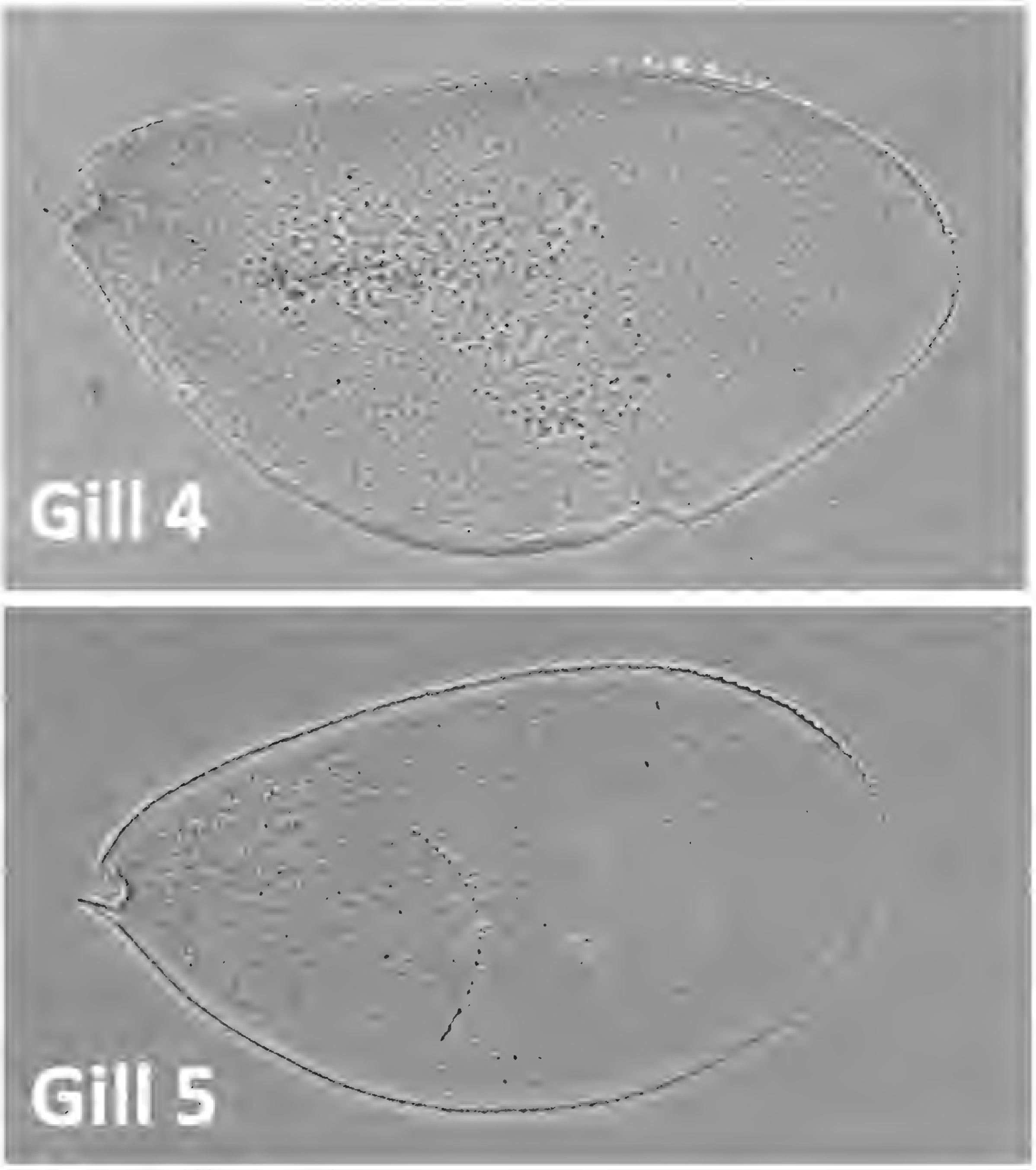

Figure 23. *Baetis vernus*: gills 4 and 5.

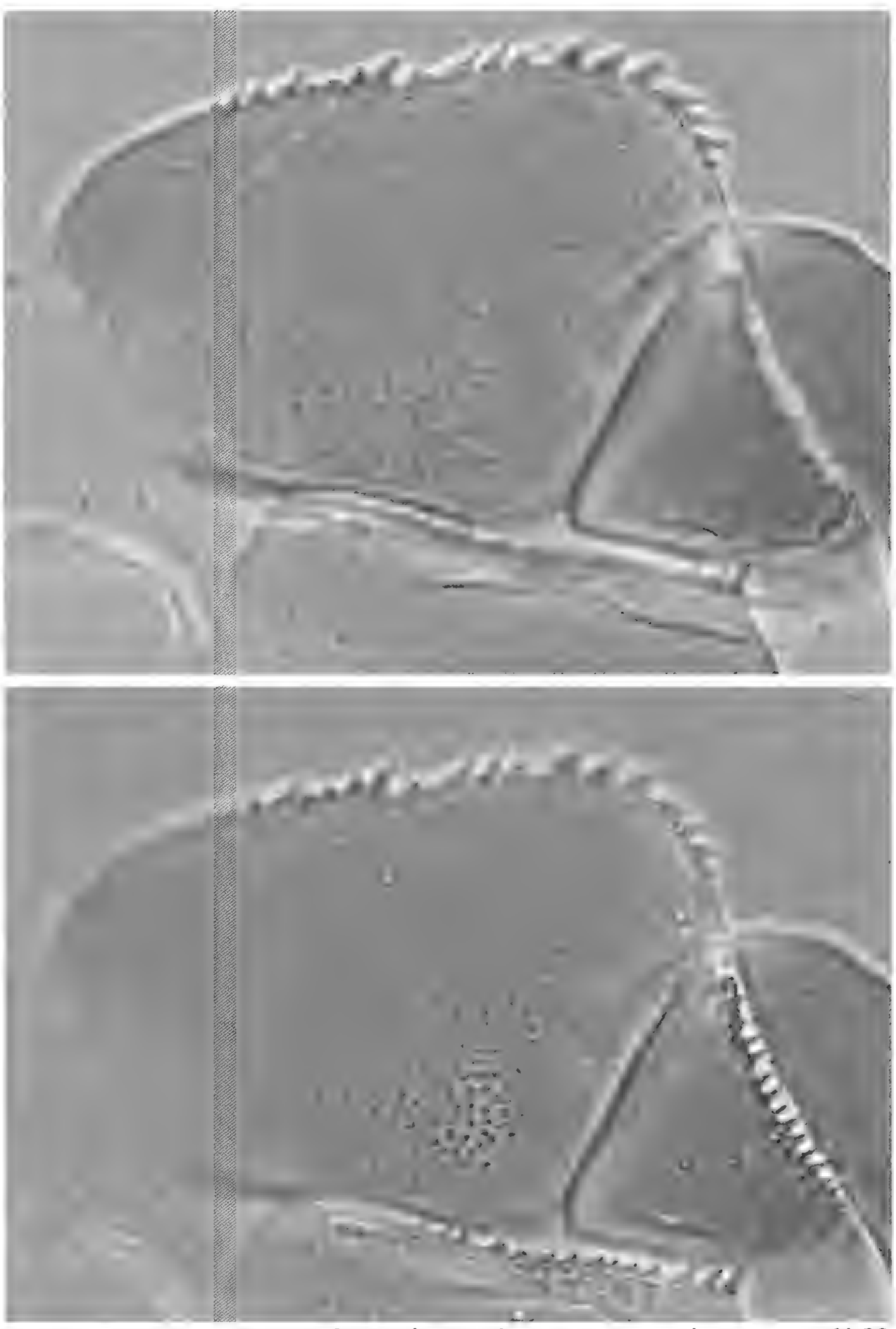

Figure 24. *Baetis vernus*: paraprocts, showing the same view at different focus settings to show setal patterns.

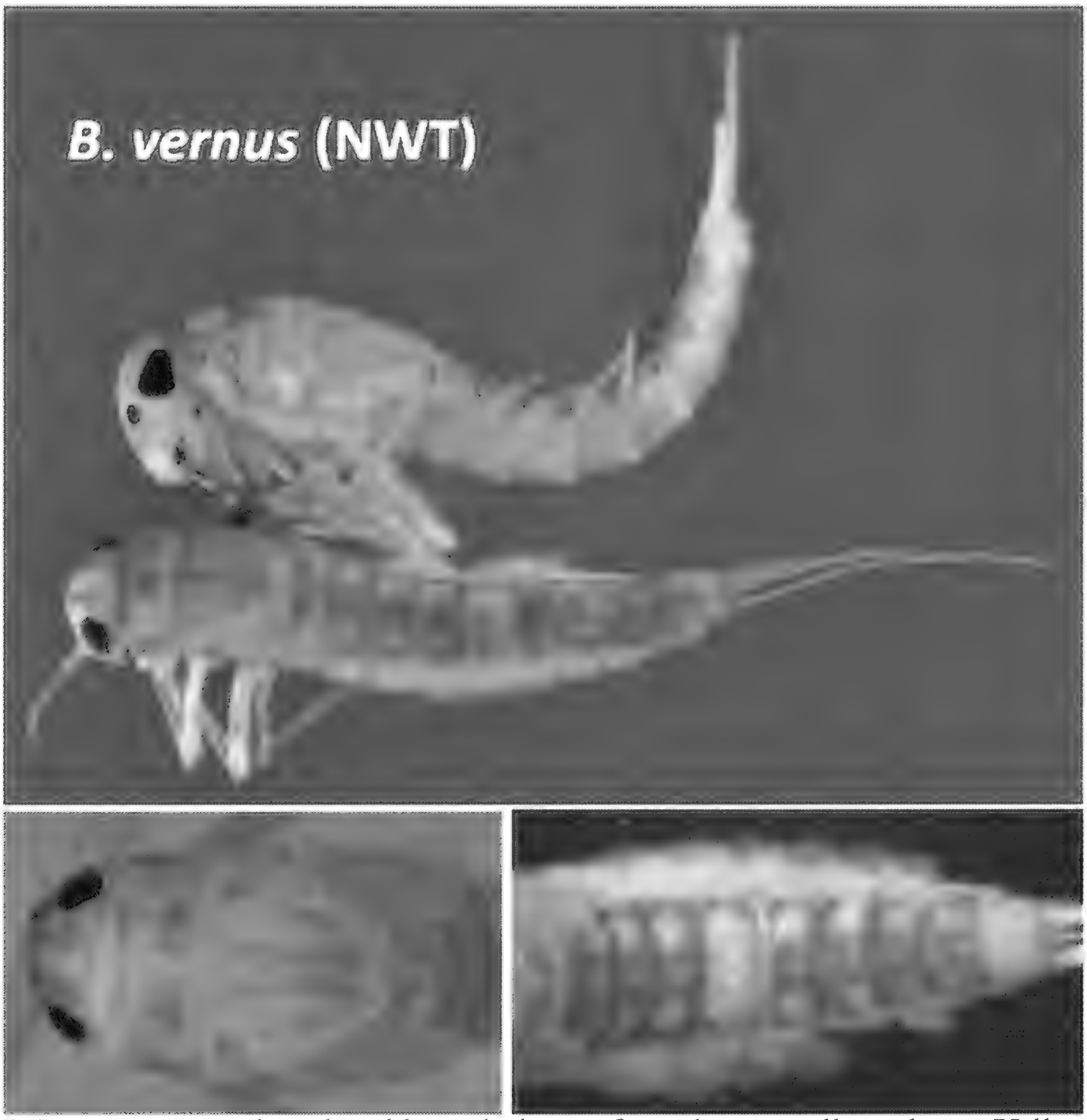

Figure 25. *Baetis vernus*: dorsal and lateral views of two larvae collected near Yellowknife in Northwest Territories, showing colour patterns and body shape.

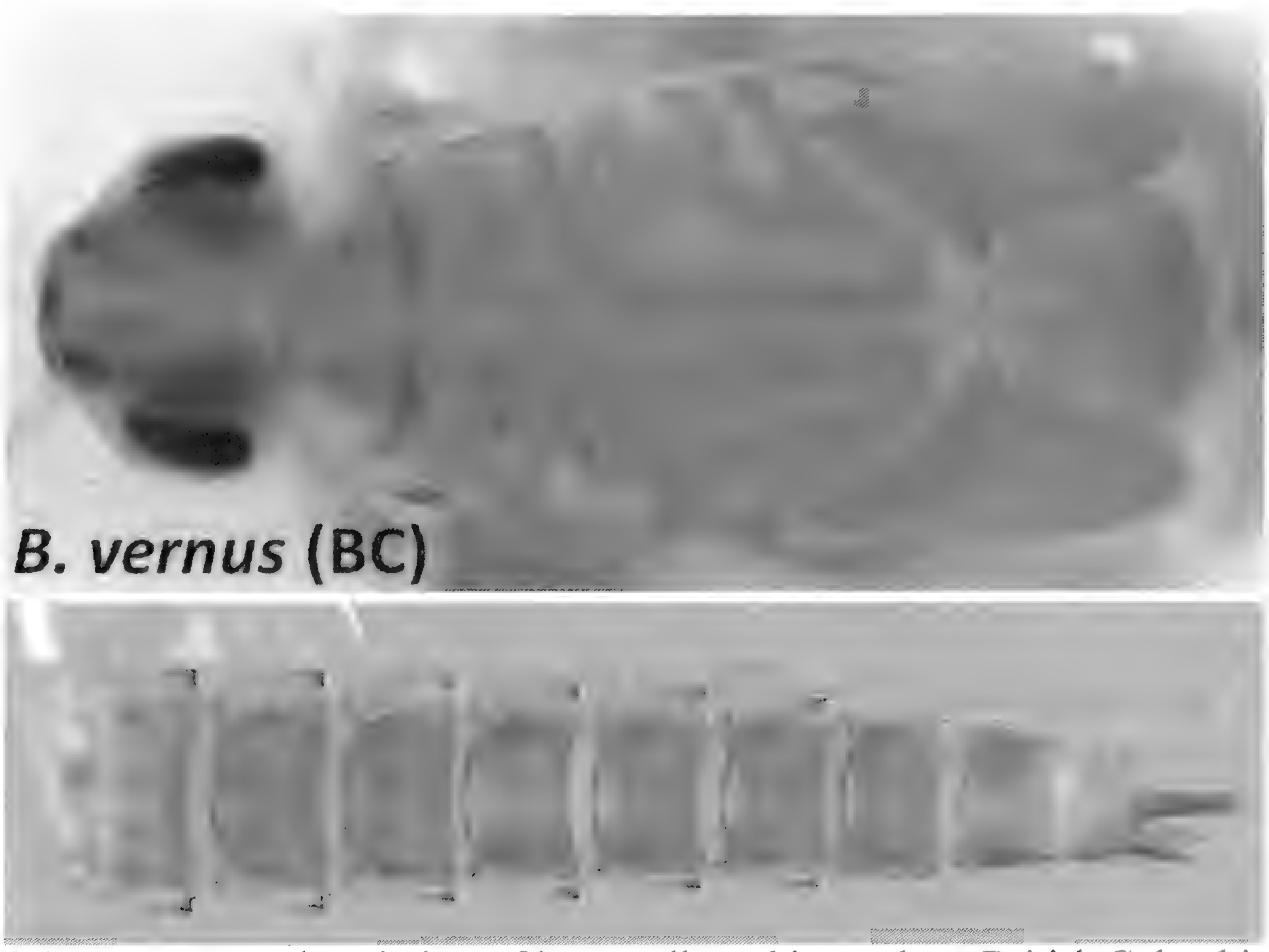

Figure 26. *Baetis vernus*: dorsal view of larva collected in northern British Columbia, showing colour patterns and body shape.

Diagnosis of larvae of *Baetis vernus* group species in North America:

In North America, the *Baetis vernus* group includes: *B. brunneicolor*, *B. bundyae*, *B. hudsonicus*, and *B. vernus*. Larvae of *B. bundyae* and *B. hudsonicus* can be separated from those of *B. brunneicolor* and *B. vernus* by the elongate abdominal gills, which are distinctly longer than twice their width. *Baetis bundyae* can be separated from *B. hudsonicus* by the presence of a short terminal filament, usually much shorter than lengths of adjacent cerci, whereas *B. hudsonicus* has a long terminal filament that is about equal to the length of adjacent cerci (secondarily, populations of *B. hudsonicus* seem to be completely parthenogenetic, no males have been detected).

Larvae of *Baetis vernus* can be separated from those of *B. brunneicolor* by the presence of the following combination of characters:
(1) Dorsal surface of body with distinctive contrasting colour pattern of brown with large pale spots and marks (Fig. 27), especially on abdominal terga, terga V and X mostly pale but other terga brown with large paired pale submedian spots,
(2) General shape of abdomen from dorsal perspective somewhat cylindrical appearing to very gradually taper from segments I to X,
(3) Dorsal surface of labrum with subtriangular raised area flanked by two brown bands that converge medially near base of notch in anterior margin, dorsal setal formula 1+3–4,
(4) Prostheca of right mandible with cluster of setae along inner edge near apex,
(5) Paraglossae of labium with apices distinctly curved inward,
(6) Femora with large blunt setae along outer edge with narrow bases and broad ends,
(7) Foretibia and -tarsus slender,
(8) Foreclaw with 8–10 denticles that gradually enlarge from base of claw toward tip,
(9) Tip of claw not attenuated (i.e., narrowed) beyond denticles,
(10) Abdominal terga with distinctive dark brown shading around gill insertions and spinules along posterior margins dark brown,
(11) Cuticle of abdominal terga moderately grainy with many distinct cuticular ridges among bases of scale setae, and
(12) Abdominal gills 2–4 with only faint traces of the medial trachea and trachea not visible on other gills.

Mature larvae of *B. brunneicolor* can usually be separated from those of *B. vernus* by the presence of the following combination of characters:
(1) Dorsal surface of body relatively uniformly brown, lacking large distinct contrasting pale spots or marks, especially on abdominal terga where some small paired dark marks are present (Fig. 27), Tergite X is usually a uniform light brown on mid-instar larvae but can be pale on black wing pad larvae,
(2) General shape of abdomen from dorsal perspective conical appearing to taper more distinctly from segments I to X,
(3) Dorsal surface of labrum with rounded raised area with no associated dark bands, dorsal setal formula 1+4–5,
(4) Prostheca of right mandible with single row of uniformly spaced setae along inner edge near apex,
(5) Paraglossae of labium with apices straight or only slightly curving inward,
(6) Femora with large blunt setae that usually have uniform width from base to tip,
(7) Foretibia and -tarsus stout,
(8) Foreleg claw with only about nine denticles that appear to change more abruptly in size from base of claw toward tip,
(9) Tip of claw attenuated (i.e., narrowed) beyond denticles,
(10) Abdominal terga with only small areas of brown shading around gill insertions and spinules along posterior margins not darker that rest of surface,
(11) Cuticle of abdominal terga weakly grainy with few widely spaced cuticular ridges among bases of scale setae, and
(12) Abdominal gills 2–6 with distinct medial trachea, lateral trachea also visible on larger gills.

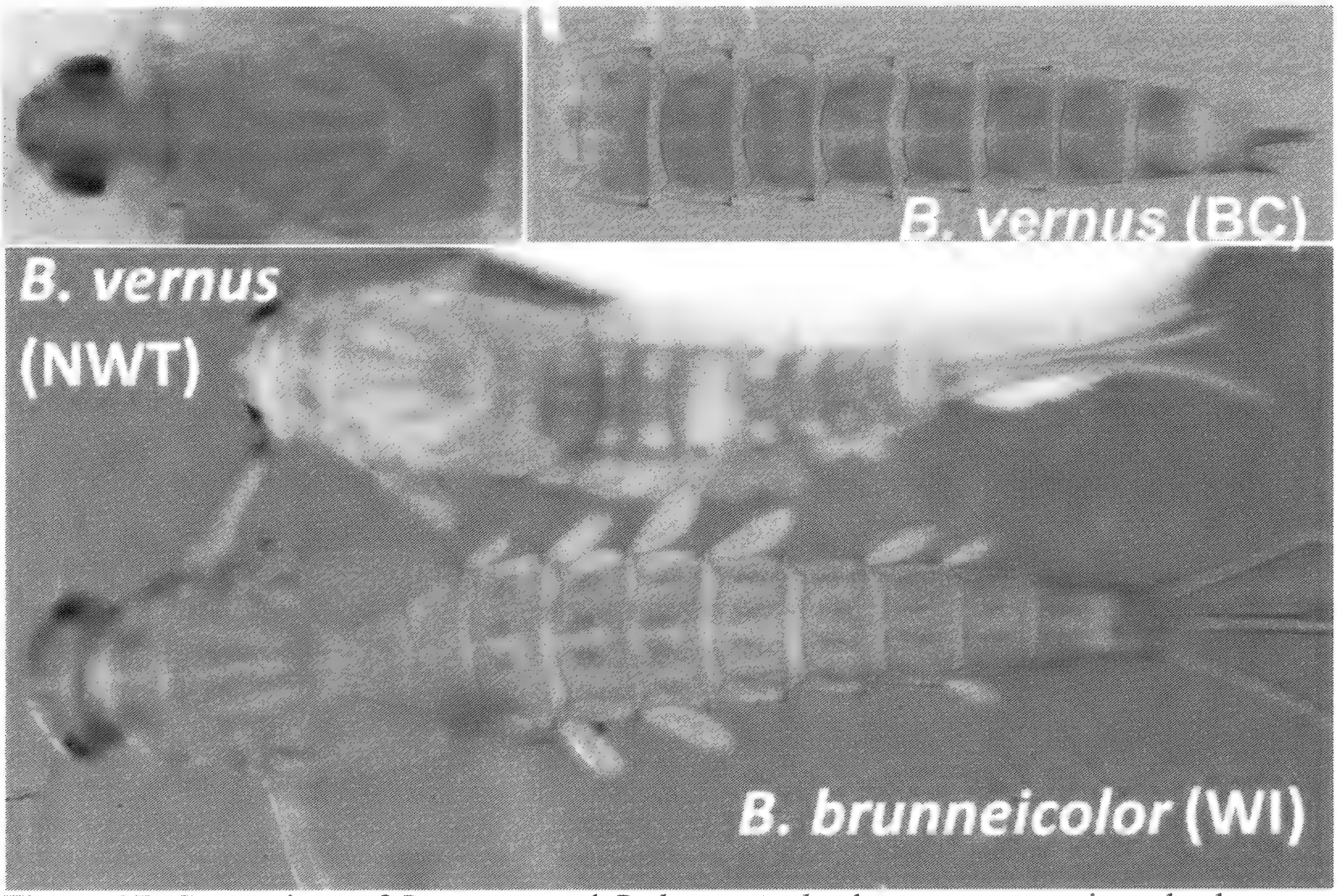

Figure 27. Comparison of *B. vernus* and *B. brunneicolor* larvae at approximately the same stage of development. The *B. vernus* specimens were collected from near Yellowknife in Northwest Territories and from the Crooked River in northern British Columbia, and the *B. brunneicolor* larva is from Wisconsin.

Distribution of *B. vernus* in Canada

The distribution of *B.* vernus in Canada is still unclear, as there are currently only four verified specimens of *B. vernus* from North America. Their distribution ranges from central British Columbia to the south–central Northwest Territories (Fig. 28, Table 1). However, *B. vernus* overlaps in distribution with *B. brunneicolor* (Fig. 28), so some specimens previously identified as *B. brunneicolor* from this region may be *B. vernus*, which could extend the distribution considerably. The specimens mapped in Fig. 4 (and listed in Table 1) were confirmed through comparison with confirmed specimens of *B. brunneicolor* and descriptions of *B. vernus* from the Palearctic. Another potential source of confusion stems from a group of specimens from northern Yukon and the Mackenzie Mountains that show characters of both species and may represent hybrid forms or a new species in the *B. vernus* group (Table 1). We show that a combination of DNA barcoding and morphological examination can resolve the two species, but targeted collecting should occur through northern Canada to obtain specimens for molecular examination to assess distribution patterns for species within the entire *Baetis vernus* group.

Phylogeography

Morphological analyses and DNA barcoding now confirm the Nearctic presence of *B. vernus*, and the locations of the currently known specimens of *B. vernus* indicate a widespread distribution in North America. Such a Holarctic distribution is not surprising as it is analogous to the distribution of *B. bundyae* (Giberson *et al.* 2007; Savolainen *et al.* 2014) and other Ephemeroptera (Kjærstad *et al.* 2012) and because *Baetis* spp. may be particularly pre-adapted for rapid dispersal due to their wing-loading characteristics (Corkum 1987). These results may also imply a widespread northern Asian distribution for *B. vernus* – or a common ancestor of it and other members of the *B. vernus* group – leading to a Beringian dispersal event. Members of the *B. vernus* group seem to be variably tolerant of a range of lentic (standing water) and lotic (running water) habitats

(Bauernfeind and Humpesch 2001; Giberson *et al.* 2007; Savolainen *et al.* 2007; Drotz *et al.* 2012), and differential use of such habitats may be a driver of structured populations or speciation (Drotz *et al.* 2012; Ståhls and Savolainen 2008). From our direct knowledge of collections (Cordero *et al.* 2017; Huber *et al.* 2019) or extrapolation from GPS coordinates (BOLD specimens in this study), the North American *B. vernus* specimens were collected in a range of situations, including a marshy area (Cordero *et al.* 2017), a slow-moving outflow of a lake (Huber *et al.* 2019), and seemingly typical lotic environments (BOLD specimens). *Baetis vernus* in Europe is mostly – but not exclusively – known from lotic systems (Savolainen *et al.* 2007). This seeming ability to reproduce and survive in a variety of habitats may have also aided *B. vernus*' dispersal ability.

DNA barcoding was vital for the initial detection of this species in North America and remains a valuable tool for distinguishing between *B. brunneicolor* and *B. vernus* (as well as potential new species in the group) in northern Canada. Morphological work on these specimens has revealed new questions regarding *B. vernus* group taxonomy and phylogeograhy, and these results highlight the need for substantial further collection of the *B. vernus* group in northern Canada. The growing use of eDNA surveys of likely habitat will be important for extending our knowledge of this and other mayfly species.

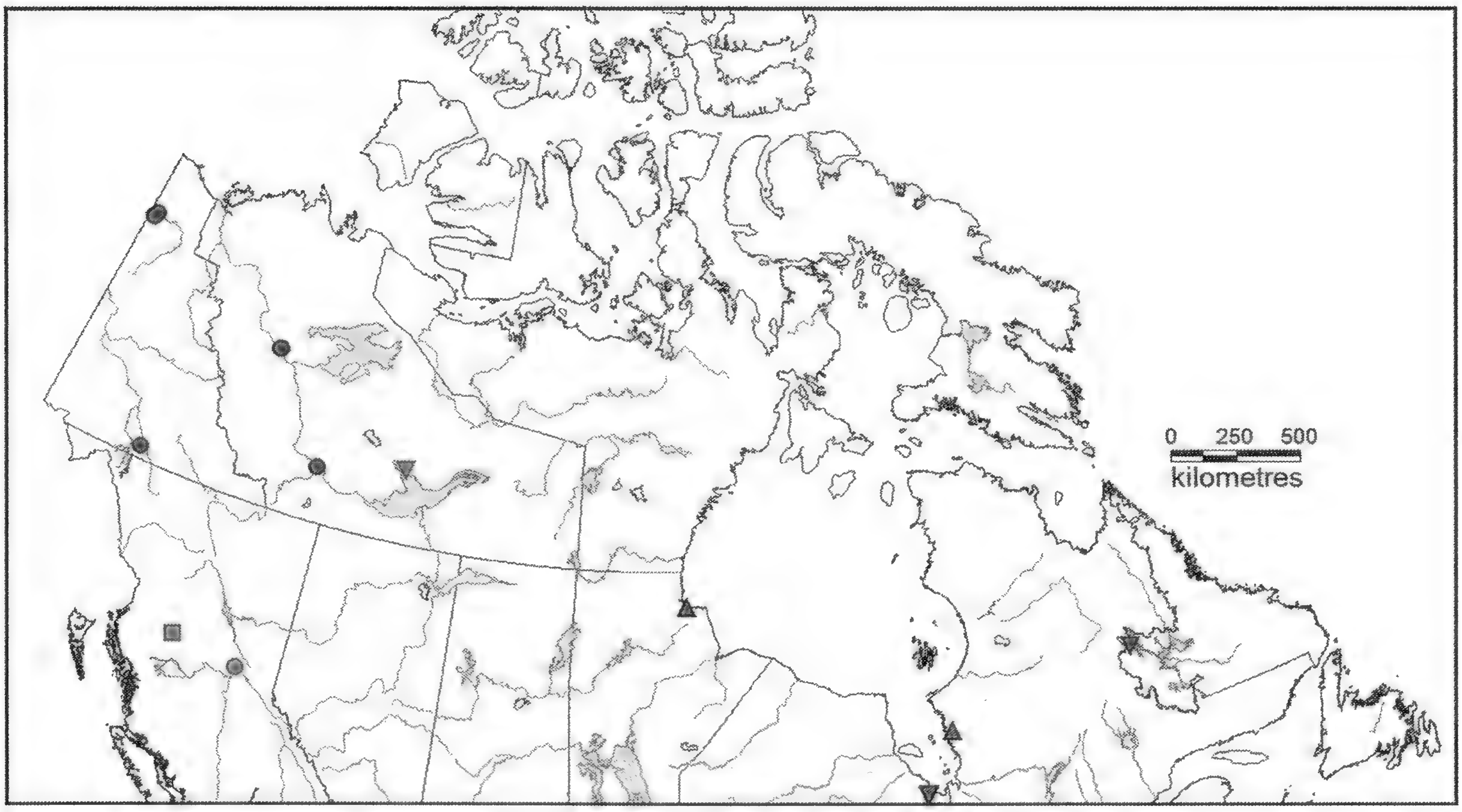

Figure 28: Confirmed record locations for *B. vernus* and *B. brunneicolor* across northern Canada. Red symbols = *B. vernus*, with different symbol shapes denoting different collections [red inverted triangle: Cordero *et al.* (2017); red circle: Huber *et al.* 2019; red square: BOLD-mined *B. vernus* data (two specimens)]. Blue symbols = *B. brunneicolor*, with different symbol shapes denoting different collections [blue circles: Giberson and Burian (2017), confirmed through comparison with eastern *B. brunneicolor* specimens; blue inverted triangles: Cordero *et al.* (2017); blue triangles: adult specimens reported in Harper and Harper 1981].

ACKNOWLEDGEMENTS

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Corrections for the Hemiptera: Heteroptera of Canada and Alaska

G.G.E. SCUDDER[1]

ABSTRACT

A total of 175 changes to the current checklist of Hemiptera: Heteroptera of Canada and Alaska are reported. Eighty deletions, eighty-eight nomenclature changes, and seven spelling corrections are detailed. In addition, comments are given on *Anthocoris tomentosus* Péricart, *Orius diespeter* Herring, *O. tristicolor* (White), and *Tupiocoris agilis* (Uhler).

Key words: Changes, checklist, Heteroptera, Canada, Alaska

INTRODUCTION

Maw *et al.* (2000) published a checklist of the Hemiptera of Canada and Alaska, giving details of the occurrence of the species of Heteroptera. Since then, there have been a large number of taxonomic changes that have resulted in deletions and nomenclature modifications for many of the taxa. In addition, a few spelling errors have been noted. Details of the 175 changes are outlined here, and comments on four taxa are given.

The order of taxa follows Maw *et al.* (2000), but species are listed in alphabetical order in each family.

Museum abbreviations are as follows:

CNC	Canadian National Collection of Insects, Agriculture and Agri-Food Canada, Ottawa, Ontario
RBCM	Royal British Columbia Museum, Victoria, B.C.
UAM	University of Alaska Museum, Fairbanks, Alaska.
UBCZ	Spencer Entomological Collection, Beaty Biodiversity Museum (formerly Spencer Entomological Museum, Department of Zoology) University of British Columbia, Vancouver, B.C.
USNM	National Museum of Natural History (formerly United States National Museum), Washington, D.C.

SYSTEMATIC TREATMENT

I. Deletions

Family CORIXIDAE

Glaenocorisa quadrata Walley

This corixid was originally described by Walley (1930) from Quebec. Jaczewski and Lansbury (1961) followed Ossianilsson (1960) and considered *G. quadrata* a synonym of *G. cavifrons* (Thomson), and stated that *G. cavifrons* was at most a subspecies of *G. propinqua* (Fieber). Although doubted by Brown (1946), this was accepted by Jansson (1986), who concluded that there were two subspecies of *G. propinqua*, with *G. propinqua cavifrons* occurring in North America. However, as noted by Jansson (2002), *G. cavifrons* was raised to specific status by Jansson (2000), because the two subspecies are sympatric in Scotland and northern Finland. Hence, *G. quadrata* Walley should be

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deleted, and all occurrence records in North America placed under *G. cavifrons* (Thomson).

Sigara modesta (Abbott)

This species was recorded from British Columbia by Downes (1934) as *Arctocorisa modesta*, with a listing of material from Vernon, 26.ix.1919 (W. Downes), determined by G.S. Walley. This record was accepted by Polhemus *et al.* (1988) and repeated in Maw *et al.* (2000).

Sigara modesta was not listed from British Columbia by Hungerford (1948), Lansbury (1960), or Scudder (1977). Scudder (1977) excluded *S. modesta* (Abbott) from the British Columbia list of Corixidae, noting that there were no other records of this species in the province. He also noted that other specimens in the Downes collection that were labelled "*modesta*" were in fact *S. grossolineata* Hungerford and that the Vernon determination was probably incorrect. Unfortunately, he overlooked the Vernon record of *S. washingtonensis* listed in Hungerford (1948), even though there were specimens with the appropriate date in the Downes collection that had been donated to the UBCZ collection in 1958. However, Scudder (1977) did quote a Vernon record in Lansbury (1955), although the date in the latter was printed as 26.ix.1929 (W. Downes).

Hungerford (1948) lists a male specimen with data Vernon, 26.ix.1919 (W. Downes), under his new species *S. washingtonensis*. In the UBCZ collection, with collection numbers COR3139-COR3141, I have located one male and three females with data 'Vernon, 26.ix.1919 (W. Downes)'. These are from the Downes collection donated to UBCZ in 1958; it is assumed that these are the specimens mentioned by Downes (1934). Hence, the record of *S. modesta* (Abbott) from British Columbia should be deleted.

Trichocorixa verticalis fenestrata (Walley)

This is treated the same as *T. verticalis verticalis* (Fieber) by Jansson (2002). Hence, *T. verticalis fenestrata* should be deleted and records should be placed under *T. verticalis verticalis*.

Family SALDIDAE

Salda anthracina Uhler

This saldid was recorded from Alaska and British Columbia by Polhemus (1988) and from Alaska, British Columbia, Northwest Territories, and the Yukon Territory in Maw *et al.* (2000). Specimens in the UBCZ collection from Alaska, Northwest Territories, and the Yukon were initially determined by me as *S. anthracina* using the key in Schuh (1967), with particular attention paid to the fact that this key noted that the second antennal segment in *S. anthracina* was pale. At that time, I had not seen specimens of *S. anthracina* from elsewhere. This led me to record *S. anthracina* in Maw *et al.* (2000). These specimens were as follows:

AK: Donnelly Cr., Richardson Hwy., 15.vii.1985 (S.G. Cannings) [UBCZ].
NT: 15 km N of BC border, Liard Hwy., 26.vi.1985 (E. Bijdemast) [UBCZ].
YK: Dawson, 31 km E, 26.vi.1980 (Bruce Gill) [UBCZ].
Kluane N.P., Slims River flats, 21.vii.1979 (G.G.E. Scudder) [UBCZ].
Kluane, Slims R. delta, 7.viii.1986, (S.G. Cannings) [UBCZ].
Mi 1059 Alaska Hwy., Kluane L., 5.vii.168 (Campbell-Smetana) [CNC].
Moose Cr., 68°31'N 137°01'W, 26.vii.1982 (G.G.E. Scudder) [UBCZ].
Von Wilczek L., 2.vii.1980 (Bruce Gill) [UBCZ].

After receiving one male and one female determined by the late J.T. Polhemus as *S. provancheri* Kelton & Lattin, and noting that these specimens from Colorado, Weld County, Pawne National Grasslands, July 1970 (R.T. Bell), had the second antennal segment mostly pale and the first segment dorsally flavescent and ventrally fuscous, I redetermined my western specimens as *S. provancheri* and not *S. anthracina.* As a result, in Scudder (1997), I recorded *S. provancheri* from Dawson (31 km E), Moose Cr., Slims R. delta and von Wilczek Lks. The specimens from Alaska and the Northwest Territories were also determined as *S. provancheri*, and not *S. anthracina*.

I note that Schuh (1967) stated that *S. anthracina* is quite variable morphologically and lives in situations similar to those preferred by S. *provancheri*, which was recorded by Schuh (1967) as *S. bouchervillei*. I have been unable to trace the original record of *S. anthracina* from Alaska, although this record is reported by Drake and Hoberlandt (1950), Drake and Hottes (1950), and Drake (1952). D. Sikes (in litt., 15 March 2018) informs me that there are no specimens under *S. anthracina* in the University of Alaska Museum.

All the specimens I have seen from Alaska that might be S. *anthracina* are, in fact, *S. provancheri*.

Drake and Hottes (1950) cite a record of *S. provancheri* as *S. bouchervillei*, from Alaska (Rampart), noting that his species is quite variable in size and degree of wing development. *Salda provancheri* was also recorded from Cook Inlet, Valdez Bay, in Alaska by Bahr and Schulte (1976). Polhemus (1988) recorded *S. provancheri* from Alaska, British Columbia, and the Northwest Territories.

Salda anthracina was recorded from British Columbia by Downes (1927) as *Lampracanthia anthracina*, with the observation that the British Columbia material was in the CNC. I have been unable to locate specimens of *S. anthracina* from British Columbia in the CNC, and this absence has been confirmed by H.E.L. Maw (in litt., 22 Feb. 2018). However, there are specimens of *S. provancheri* from British Columbia in the CNC, RBCM, and UBCZ collections, with some of the latter being recorded by Downes (1927) as *S. coriacea*, a synonym of *S. provancheri*.

Hence, it is evident that the records of *S. anthracina* from British Columbia, the Northwest Territories, and the Yukon in Maw *et al.* (2000) should be deleted. The occurrence of *S. anthracina* in Alaska needs to be confirmed.

Family ANTHOCORIDAE

Tetraphleps uniformis Parshley

Lattin (2006) has shown that *T. uniformis* Parshley is a synonym of *T. canadensis* Provancher, and restored *T. furvus* Van Duzee as a valid species in its place. Hence, *T. uniformis* should be deleted and replaced by *T. furvus* Van Duzee.

Xylocoris umbrinus Van Duzee

Lattin (2005) has shown that *X. umbrinus* Van Duzee is a synonym of *X. californicus* (Reuter). Thus, *X. umbrinus* Van Duzee should be deleted and replaced by *X. californicus* (Reuter).

Family NABIDAE

Pagasa fusca (Stein)

After Kerzhner (1993a) raised *P. nigripes* Harris to specific status and recorded this species from Alberta, Saskatchewan, Quebec and Alaska, Scudder (2008) showed that all the specimens of *P. fusca* (Stein) reported from the Yukon and the Northwest Territories, and some specimens from British Columbia, were *P. nigripes*. Thus, *P. fusca* should be deleted from the Yukon and Northwest Territories.

Family MIRIDAE

Adelphocoris superbus (Uhler)

Schwartz and Scudder (2003) concluded that *A. superbus* (Uhler) is a synonym of *A. rapidus* (Say). Hence, *A. superbus* and all three provincial records should be deleted.

Agnocoris pulverulentus (Uhler)

This species was first reported from Alaska (Fort Yukon) by Moore (1955). Moore (1956) did not list the Alaska (Fort Yukon) material when he recorded *A. rubicundus* (Fallén) in the New World, but considered this latter species as Holarctic. Wheeler and Henry (1992) also did not record *A. rubicundus* from Alaska, although they stated that this species may have survived in an Alaska refugium. Maw *et al.* (2000), while recording A. *pulverulentus* in Alaska following Moore (1955), also noted *A. rubicundus*

from Alaska. This was based on specimens from Alaska in the CNC determined by M.D. Schwartz as *A. rubicundus*. Included was material labelled 'Alaska, Fort Yukon, 900', 4.viii.1951 (H.C. Severin)'. T.J. Henry informs me (in litt., 15 February 2018), that he could not locate Alaska specimens of *Agnocoris* in the USNM, although Moore (1955) recorded one male and three females as *A. pulverulentus* from Alaska, Fort Yukon, July 18, 1951 (R.I. Sailer) [USNM].

As a result, I hereby delete the record of A. *pulverulentus* from Alaska, assuming it is in fact *A. rubicundus*.

Aoplonema uhleri (Van Duzee)

Forero (2008) has shown that *Hadronema uhleri* Van Duzee is a synonym of *A. princeps* (Uhler). *Aoplonema uhleri* should be deleted and replaced by *A. rubrum* Forero.

Capsus ater (Linnaeus)

This species was reported from Alberta (Edmonton) by Blatchley (1926), quoted from Alberta by Henry and Wheeler (1988), and reported by Maw *et al.* (2000). The record was doubted by Wheeler and Henry (1992), and it was not included from Alberta in Kelton (1980). All the specimens of *Capsus* that I have examined from Alberta are *C. cinctus* (Kolenati). This latter species, recorded as *C. simulans* (Stål), was first reported from Banff and Lethbridge in Alberta by Knight (1926) and subsequently were recorded under this name from Alberta by Strickland (1953), MacNay (1953), and Kelton (1980).

Hence, it is assumed that the record of *C. ater* from Alberta should be deleted. Vinokurov (1977) synonymized *C. simulans* (Stål) with C. *cinctus* (Kolenati) and noted that this species occurred in North America from Alaska to Iowa in the United States.

Chlamydatus pullus (Reuter)

Many of the records formerly placed under *C. pullus* (Reuter) by Kelton (1965), Scudder (1997), and Maw *et al.* (2000) are now placed under the species *C. keltoni* Schuh & Schwartz (Schuh and Schwartz 2005). *Chlamydatus pullus* (Reuter) as noted by Schuh and Schwartz (2005) is found only in Quebec, Saskatchewan, and the Yukon. The latter were recorded as "*Chlamydatus* sp. near *auratus* Kelton" by Scudder (1997).

The result is that all records of *C. pullus* in Canada, except those from Quebec, Saskatchewan, and the Yukon, should be deleted.

Coquillettia insignis (Uhler)

Wyniger (2011) revised the genus *Coquillettia* Uhler and found that *C. insignis* Uhler is confined to California. The species in Alberta and Saskatchewan was described as a new species *C. schwartzi* Wyniger and the specimens from British Columbia as being either of two new species, described as *C. pergrandis* Wyniger or *C. thomasi* Wyniger.

Hence, *C. insignis* Uhler, in Maw *et al.* (2000), should be deleted and replaced by the species listed above.

Dacota hesperia Uhler

This species was recorded from British Columbia in Maw *et al.* (2000), based on a single female specimen from B.C., Fraser, 29.vii.1982 (G.G.E. Scudder). This specimen was subsequently determined in 2010 by M.D. Schwartz as *Pinophylus rolfsi* (Knight) and recorded as AMNH_PBI00394201. *Pinophylus rolfsi* is now *P. alpinus* (Van Duzee), according to Schwartz (2013).

Thus, the record of *D. hesperia* Uhler from British Columbia should be deleted.

Dicyphus vestitus Uhler

Dicyphus vestitus was recorded from British Columbia by Parshley (1919) and Downes (1927). Parshley (1919) cited specimens from B.C., Saanich Dist., V.I., Apr. 30, Sept. 14, 1918 (W. Downes), and Downes (1927) cited specimens from Goldstream, Sept. 9th, 1923 (K.F. Auden), Vernon, May 6th, 1920 (H.R. Ruhmann), and Victoria, Sept. 7th, 1920 (W. Downes).

Based on these records, *D. vestitus* was recorded from British Columbia by Henry and Wheeler (1988), and this record was repeated by Maw *et al.* (2000).

In the UBCZ collection, which now contains the late W. Downes collection, there are specimens of *D. discrepans* Knight that are labelled B.C., Saanich Dist., 14.ix.1918 (W.

Downes) and B.C., Goldstream, 9.ix.1923 (K.F. Auden): these are evidently specimens listed by Parshley (1919) and Downes (1927), respectively. Although Knight (1923) described *D. discrepans* and distinguished it from *D. vestitus*, *D. discrepans* was not listed by Downes (1927). It is evident that the early records of *D. vestitus* in Parshley (1919) and Downes (1927) should be assigned to *D. discrepans*.

Hence, the *D. vestitus* Uhler record from British Columbia in Maw *et al.* (2000) should be deleted. Henry (1999a) gives a recent key to *D. discrepans* and *D. vestitus*.

Lopidea confluenta (Say)

Maw *et al.* (2000) recorded *L. confluenta* (Say) from Alberta, Manitoba, Ontario, and Quebec.

Lopidea confluenta is not recorded from Alberta by Strickland (1953) nor from the prairie provinces by Kelton (1980). However, it is listed from Ontario and Quebec by Asquith (1991) and Wheeler and Henry (1988). It was recorded from Manitoba (Aweme) by Criddle (1921), and this was the basis for its inclusion in Scudder (2014).

It is evident that the Alberta record is an error and should be deleted. The Manitoba record needs to be confirmed.

Lopidea nigridea serica Knight

This was reported from Alaska in Maw *et al.* (2000), based on one female specimen from Tok, 22.vii.1982 (L.A. Kelton) [CNC]. However, M.D. Schwartz has since determined that this specimen is *L. dakota* (Knight).

Hence, *L. nigridea serica* Knight should be deleted for Alaska, as noted by Scudder and Sikes (2014).

Megalopsallus lycii (Knight)

Europiella lycii Knight 1968 was transferred to the genus *Megalopsallus* Knight by Schuh *et al.* (1995) and synonymized with *M. humeralis* (Van Duzee) by Schuh (2000). The latter species does not occur in Canada and should therefore be deleted. The Alberta and Saskatchewan records under *M. lycii* in Maw *et al.* (2000) should be assigned to *M. sparsus* (Van Duzee) (Schuh 2000).

Megalopsallus montanae (Knight)

Europiella montanae Knight 1968 was transferred to the genus *Megalopsallus* Knight by Schuh *et al.* (1995) and synonymized with *M. nigrofemeratus* (Knight) by Schuh (2000). Hence, *M. montanae* (Knight) can be deleted.

Melanotrichus concolor (Kirschbaum)

This European species was reported from Quebec as *Orthotylus concolor (*Kirschbaum) by Moore (1980), Larochelle (1984), and Roch (2008). This record as *M. concolor* (Kirschbaum) was reported from Quebec by Henry and Wheeler (1988) and repeated by Maw *et al.* (2000).

However, Henry (1991) could not confirm the identity of Quebec specimens and believed that they actually are *M. virescens* (Douglas & Scott). As a result, the record of *M. concolor* from Quebec should be deleted and replaced by *M. virescens*.

Microphylellus elongatus Knight

Microphylellus elongatus Knight was cited as a synonym of *Plagiognathus flavipes* (Provancher) by Schuh (2001) (see below). Hence, *M. elongatus* Knight should be deleted.

Orectoderus salicis Knight

This species has been synonymized with *O. montanus* Knight by Wyniger (2010). Hence, it can be deleted.

Orthotylus candidatus Van Duzee

Scudder (2008) reported that the earlier records of *O. candidatus* Van Duzee from Ontario and Saskatchewan were referable to *O. nyctalis* Knight. Hence, the records of *O. candidatus* Van Duzee from Ontario and Saskatchewan should be deleted.

Paradacerla downesi (Knight)

This species was recorded by Downes (1934) from B.C., Jordan Meadows on Vancouver Island, at 1700 feet (W. Downes) det Downes. However, specimens from

Jordan Meadows are not in the late W. Downes collection donated to UBCZ in 1958 and are not in the RBCM. Specimens from British Columbia were not listed in Kelton and Knight (1959), and currently *P. downesi* is unknown in British Columbia. Hence, *P. downesi* should be deleted from the Canadian list.

Pilophorus clavatus (Linnaeus)

This European species was listed from Alberta, British Columbia, Manitoba, Nova Scotia, Ontario, Quebec, and Saskatchewan by Henry and Wheeler (1988), and these records were repeated in Maw *et al.* (2000). *Pilophorus clavatus* was first reported from Newfoundland in 2005 by Wheeler *et al.* (2006).

Downes (1927) recorded *P. clavatus* determined by H.H. Knight, from British Columbia, Victoria, 17.ix.1924 (W. Downes) and Mission, 22.ix.1925 (W. Downes), while Kelton (1980) noted this species from British Columbia, Alberta, Saskatchewan, Manitoba, Ontario, and Nova Scotia. *Pilophorus clavatus* was recorded from Quebec by Moore (1950) and Larochelle (1984), but not by Roch (2008), who queried the occurrence in Ontario and New Brunswick.

Schuh and Schwartz (1988) noted that they were unable to confirm all the earlier records of *P. clavatus* in Canada, except for the records from Manitoba and Nova Scotia. Schuh and Schwartz (1988) considered that other records of *P. clavatus* in Canada could either be *P. neoclavatus* Schuh & Schwartz or misidentified other species. These comments were repeated by Wheeler and Henry (1992).

In the late W. Downes collection in the UBCZ, I found one female with the data, B.C., Victoria, 17.ix.1924 (W. Downes), which is evidently the specimen recorded by Downes (1927) from British Columbia. This specimen was determined by M.D. Schwartz in 1998 as *P. vicarius* Poppius, so the British Columbia record of *P. clavatus* should be deleted.

It would thus appear that all records of *P. clavatus* from Canada, except those for Manitoba, Nova Scotia, and Newfoundland, should be deleted.

Pilophorus uhleri Knight

This species was recorded from Alberta, British Columbia, Manitoba, Ontario, and Saskatchewan by Henry and Wheeler (1988), while Schuh and Schwartz (1988) considered *P. uhleri* an eastern North American species, occurring west to Alberta. Schuh and Schwartz (1988) gave records for Alberta, Manitoba, New Brunswick, Nova Scotia, Prince Edward Island, and Saskatchewan, but not British Columbia.

Downes (1927) reported *P. uhleri* Knight, determined by H.H. Knight, from British Columbia, Victoria, 15 Sept. 1924 (W. Downes), on *Pinus contorta*. This record was accepted by Henry and Wheeler (1988) and Maw *et al.* (2000).

However, a female specimen in the late W. Downes collection at UBCZ, with the data, B.C., Victoria, *Pinus contorta*, 15.ix.1924 (W. Downes), is evidently the specimen listed by Downes (1927). It was determined by M.D. Schwartz in 1998 as *P. americanus* Poppius.

Hence, *P. uhleri* from British Columbia, should be deleted. It may be noted that Schuh and Schwartz (1988) reported that *P. uhleri* most closely resembles *P. americanus*.

Plagiognathus albatus vittiscutis Knight

Treated as *P. albatus* (Van Duzee) by Schuh (2001). Hence, *P. albatus vittiscutis* Knight can be deleted.

Plagiognathus albonotatus Knight

Synonymized with *P. fuscosus* (Provancher) by Schuh (2001). Hence, *P. albonotatus* Knight should be deleted.

Plagiognathus caryae Knight

Synonymized with *P. albatus* (Van Duzee) by Schuh (2001). Hence, *P. caryae* Knight should be deleted.

Plagiognathus cuneatus Knight

This variety of *P. annulatus* Uhler, established by Knight (1923), was synonymized with *P. obscurus* Uhler by Schuh (2001). Hence, *P. cuneatus* Knight should be deleted.

Plagiognathus fumidus Uhler

Considered a synonym of *Europiella decolor* (Uhler) by Schuh (2001). Hence, *P. fumidus* Uhler should be deleted.

Plagiognathus fusciflavus Knight

Synonymized with *P. verticalis* (Uhler) by Schuh (2001). Hence, *P. fusciflavus* Knight should be deleted and *Plagiognathus verticalis* (Uhler) added to the B.C. listing.

Plagiognathus moerens (Reuter)

According to Schuh (2001), this species is not known to occur in Alberta and British Columbia. Records for these two provinces should be transferred to *P. shoshonea* Knight. Thus, the records of *P. moerens* (Reuter) for Alberta and British Columbia should be deleted.

Plagiognathus nigritus Knight

Synonymized with *P. brevirostris* Knight by Schuh (2001). Hence, *P. nigritus* Knight should be deleted and the Alberta record transferred to *P. brevirostris* Knight.

Plagiognathus obscurus albocuneatus Knight

Treated as *P. obscurus* Uhler by Schuh (2001). Thus, *P. obscurus albocuneatus* Knight should be deleted.

Plagiognathus politus flaveolus Knight

Treated as *P. politus* Uhler by Schuh (2001). Thus, *P. politus flaveolus* Knight should be deleted.

Plagiognathus repletus Knight

Synonymized with *P. albatus* (Van Duzee) by Schuh (2001). Hence, *P. repletus* Knight should be deleted.

Plagiognathus similis Knight

Synonymized with *P. albatus* (Van Duzee) by Schuh (2001). Hence, *P. similis* Knight should be deleted.

Psallus variabilis (Fallén)

Psallus variabilis (Fallén) was reported from Ontario by Blatchley (1926) and Henry and Wheeler (1988), and this record was repeated in Maw *et al.* (2000). However, Wheeler and Henry (1992) reported that this Ontario record was incorrect, Knight (1927) having noted that early records of *P. variabilis* in North America were incorrect and that specimens were misidentified. Knight (1927) said that these early records refer to *Lepidopsallus rubidus* var *atricolor* Knight, which Wheeler and Henry (1992) call *Atractotomus atricolor* (Knight). However, Stonedahl (1990) does not record *A. atricolor* (Knight) from Ontario, although Stonedahl (1990) reported *A. rubidus* (Uhler) from Ontario. Nevertheless, valid records for *P. variabilis* (Fallén) in North America were given by Wheeler and Hoebeke (1982) and Wheeler and Henry (1992): these did not include Ontario. Larochelle (1984) synonymized *L. rubidus* var *atricolor* Knight with *L. rubidus* (Uhler).

It is evident that the record of *P. variabilis* (Fallén) from Ontario and Canada in Maw *et al.* (2000) should be deleted.

Sixeonotus insignis Reuter

This species was recorded from Quebec by Larochelle (1984). However, Quebec specimens from Knowlton, 4.vii.1929 (G.S. Walley), Knowlton, 8.viii.1929 (L.J. Milne), and Otter Lake, 24.vii.1958 (L.A. Kelton) in the CNC have been determined by M.D. Schwartz in 2000 as *S. deflatus* Knight. Hence, the Larochelle (1984) record from Quebec probably refers to *S. deflatus*. Thus, the *S. insignis* Reuter record from Quebec should be deleted.

Slaterocoris robustus (Uhler)

This species was recorded from Alberta in Maw *et al.* (2000), but it was not cited by Strickland (1953), Kelton (1968, 1980), Henry and Wheeler (1988), or Schwartz (2011). Evidently, this record for Alberta was a mistake and should be deleted. The record of *S. robustus* (Uhler) from British Columbia was confirmed by Schwartz (2011).

Trigonotylus americanus Carvalho

In the original description of *T. americanus*, in Carvalho and Wagner (1957), paratypes were listed from British Columbia, Vernon, vii-i-47 (H.B. Leech). Based on determinations by the late L.A. Kelton, *T. americanus* was recorded from Alaska (Hope) and the Yukon by Scudder (1997), and so recorded in Maw *et al.* (2000). As noted by Scudder and Sikes (2014), a male specimen with the data 'Alaska, Hope, Kenai Pen., 15.ii.1951 (W.J. Brown)' in the CNC has been determined by M.D. Schwartz as *T. viridis* (Provancher). Hence, Scudder and Sikes (2014) stated that *T. americanus* Carvalho should be removed from the list of Heteroptera from Alaska, because no other specimens of the species are known from the state.

Similarly, M.D. Schwartz has dissected males of the Yukon specimens listed as *T. americanus* by Scudder (1997) and found all of these to be *T. viridis* (Provancher). Golub (1989) resurrected *T. viridis* (Provancher), which Kelton (1971) considered a synonym of *T. ruficornis* (Geoffrey). All other specimens of *Trigonotylus* Fieber from the Yukon appear to be *T. viridis*. Hence, the record of *T. americanus* Carvalho from the Yukon should be deleted.

Trigonotylus tenuis Reuter

Henry and Wheeler (1988) reported *T. doddi* (Distant) from Alberta, Manitoba, and Saskatchewan. Since Golub (1989) showed that *T. doddi* was a junior synonym of *T. tenuis* Reuter, Maw *et al.* (2000) reported the Henry and Wheeler (1988) records as *T. tenuis* Reuter. However, Wheeler and Henry (1992) have noted that the original Henry and Wheeler (1988) records undoubtedly refer to other species of *Trigonotylus* Fieber. Perhaps they refer to *T. canadensis* Kelton, described from Alberta, Manitoba, and Saskatchewan by Kelton (1970).

Hence, it is reasonable to delete the record of *T. tenuis* Reuter from the prairie provinces and Canada. It is not included in Kelton (1980).

Family ARADIDAE

Aradus lugubris nigricornis Reuter

This taxon, treated as a subspecies by Froeschner (1988), was said by Parshley (1921) not to be of geographic significance, because it occurs throughout the range of the species *A. lugubris* Fallén in North America. Hence, it should be deleted.

Family ORSILLIDAE

Nysius groenlandicus (Zetterstedt)

This species in North America was recorded from Alaska, Manitoba, Newfoundland, Ontario, Prince Edward Island, and Quebec by Ashlock and Slater (1988). These records were repeated in Maw *et al.* (2000), with the addition of the Yukon and Labrador. All of these occurrence records were based on the published literature, although not all authors cited the diagnostic characters used in their identification.

The published records were: Alaska (many cited by Slater 1964); Yukon (Scudder 1997); Manitoba (Churchill) (Barber 1947a, 1947b); Newfoundland (presumably Brown 1934); Ontario (Muskoka Lake District) (Van Duzee 1889); Prince Edward Island (Barber 1947a, 1947b); Quebec (Bradore Bay) (Brown 1934; Barber 1947a; Moore 1950; Béique and Robert 1964; Larochelle 1984); Labrador (Nain) (Brown 1934).

Ashlock (1967) questioned whether *N. groenlandicus* (Zett.) occurred in North America, and this was noted by Böcher (1976). Böcher (1978) observed that *N. groenlandicus* seems to be absent in North America, and this was repeated by Danks (1981).

At present, the record of *N. groenlandicus* from Prince Edward Island should be deleted, although the identity of this material still must be determined.

Family RHYPAROCHROMIDAE

Perigenes constrictus (Say)

This species was reported from Alaska by Van Duzee (1919), and this record was repeated by Slater (1964), Ashlock and Slater (1988), Maw *et al.* (2000), and Lattin (2008). However, Scudder and Sikes (2014) noted that the female specimen in the CNC from Ketchikan, on which the Alaska record is based, is actually a specimen of *Ligyrocoris sylvestris* (Linnaeus). Hence, the record of *P. constrictus* (Say) from Alaska should be deleted, as noted by Scudder and Sikes (2014).

Scolopostethus atlanticus Horváth

This species was reported from British Columbia, Manitoba, Ontario, Quebec, and Newfoundland by Ashlock and Slater (1988), and these records were repeated in Maw *et al.* (2000). These provincial records were evidently based on earlier reports, namely those for Manitoba (Winnipeg) by Gibson (1912), for Ontario (Ottawa) by Gibson (1915), and for Quebec by Béique and Robert (1964); Roch (2008) also reported *S. atlanticus* from Ontario and Quebec. The records for Newfoundland were from Torre-Bueno (1917) and Slater (1964), and Torre-Bueno (1946). However, it may be noted that neither Parshley (1919) nor Downes (1927) gave records for *S. diffidens* Horváth.

Sweet (1964) gave a detailed description of the distinguishing characters of *S. atlanticus* and considered this species an eastern Nearctic taxon. He thought that most of the distribution records for *S. atlanticus* from the northern part of North America were incorrect, and he specifically noted that the records for British Columbia in Parshley (1919) referred to *S. thomsoni* Reuter. He also noted that the late H.G. Barber had frequently mistakenly named specimens of *S. thomsoni* and *S. diffidens* in the USNM as *S. atlanticus*.

I examined and photographed the male lectotype of *S. atlanticus* Horváth in Budapest in February 1965 and have not seen similar material in all the numerous specimens of *Scolopostethus* Fieber from Canada that I have examined over the past 60 years. In fact, in the late W. Downes collection donated to UBCZ in 1958, there is one short-winged female from B.C., Agassiz, 25.vii.1921 (W. Downes). This is obviously the specimen listed by Downes (1927), but it is *S. diffidens*. The same collection contains a macropterous female from B.C., Enderby, 14.x.1920 (W. Downes). This was listed by Downes (1927) as *S. atlanticus*, but is actually *S. thomsoni*. Furthermore, a short-winged female from B.C., Colquitz, 4.iv.1919 (W. Downes), and a macropterous female from B.C., Cowichan, 24.viii.1918 (W. Downes), both listed by Brown (1934) as *S. atlanticus* and now in the late W. Downes collection at UBCZ, are in fact *S. thomsoni*.

Hence, I conclude that *S. atlanticus* should be deleted from the list of species in Canada.

II. Nomenclature Changes

Family ANTHOCORIDAE

Orius minutus (Linnaeus)

Lewis and Lattin (2010) have noted that this introduced species in British Columbia is actually *O. vicinus* Ribaut. Hence, this name should be replaced with *O. vicinus*.

Family NABIDAE

Kerzhner and Henry (2008) have rearranged the checklist of the Nabidae in North America. This has resulted in a large number of nomenclatural changes. *Nabicula* Kirby and *Omanonabis* Asquith & Lattin are treated as subgenera of *Nabis* Latreille, and *Anaptus* Kerzhner is considered a subgenus of *Himacerus* Wolff. These changes result in nine nomenclatural changes in the Nabidae as follows:

- *Anaptus major* (Costa): change to *Himacerus* (*Anaptus*) *major* (Costa).
- *Nabicula* (*Dolichonabis*) *americolimbata* (Carayon): change to *Nabis* (*Dolichonabis*) *americolimbatus* (Carayon).
- *Nabicula* (*Dolichonabis*) *limbata* (Dahlbom): change to *Nabis* (*Dolichonabis*) *limbatus* Dahlbom.

- *Nabicula* (*Dolichonabis*) *nigrovittata nearctica* Kerzhner: change to *Nabis* (*Dolichonabis*) *nigrovittatus nearctica* (Kerzhner).
- *Nabicula* (*Limnonabis*) *propinqua* (Reuter): change to *Nabis* (*Limnonabis*) *propinquus* Reuter.
- *Nabicula* (*Nabicula*) *flavomarginata* (Scholtz): change to *Nabis* (*Nabicula*) *flavomarginatus* Scholtz.
- *Nabicula* (*Nabicula*) *subcoleoptrata* Kirby: change *Nabis* (*Nabicula*) *subcoleoptratus* (Kirby).
- *Nabicula* (*Nabicula*) *vanduzeei* (Kirkaldy): change to *Nabis* (*Nabicula*) *vanduzeei* (Kirkaldy).
- *Omanonabis lovetti* (Harris): change to *Nabis* (*Omanonabis*) *lovetti* Harris.

Family MIRIDAE

Coniferocoris pinicolus (Coniferocoris Schwartz & Schuh)

This genus *Coniferocoris* Schwartz & Schuh was synonymized with *Plesiodema* Reuter by Schwartz (2006). Thus, this species, listed in Maw *et al.* (2000) as *C. pinicolus*, should be changed to *Plesiodema pinicolus* (Schwartz & Schuh).

Icodema nigrolineatum (Knight)

Henry (1999b) has shown that *Plagiognathus nigrolineatum* Knight should be placed as the type species of a new genus that he named *Americodema*. Hence, the name *I. nigrolineatum* (Knight) should be changed to *Americodema nigrolineatum* (Knight).

Genus *Lygocoris*, subgenus *Neolygus* Knight

Neolygus Knight was raised to generic status by Yasunaga *et al.* (2002). This results in 29 name changes as listed below:

- *Lygocoris alni* (Knight): change to *Neolygus alni* (Knight).
- *Lygocoris atricallus* Kelton: change to *Neolygus atricallus* (Kelton).
- *Lygocoris atritylus* (Knight): change to *Neolygus atritylus* (Knight).
- *Lygocoris belfragii* (Reuter): change to *Neolygus belfragii* (Reuter).
- *Lygocoris caryae* (Knight): change to *Neolygus caryae* (Knight).
- *Lygocoris clavigenitalis* (Knight): change to *Neolygus clavigenitalis* (Knight).
- *Lygocoris communis* (Knight): change to *Neolygus communis* (Knight).
- *Lygocoris contaminatus* (Fallén): change to *Neolygus contaminatus* (Fallén).
- *Lygocoris fagi* (Knight): change to *Neolygus fagi* (Knight).
- *Lygocoris geneseensis* (Knight): change to *Neolygus geneseensis* (Knight).
- *Lygocoris hirticulus* (Van Duzee): change to *Neolygus hirticulus* (Van Duzee).
- *Lygocoris inconspicuus* (Knight): change to *Neolygus inconspicuus* (Knight).
- *Lygocoris invitus* (Say): change to *Neolygus invitus* (Say).
- *Lygocoris johnsoni* (Knight): change to *Neolygus johnsoni* (Knight).
- *Lygocoris knighti* Kelton: change to *Neolygus knighti* (Kelton).
- *Lygocoris laureae* (Knight): change to *Neolygus laureae* (Knight).
- *Lygocoris omnivagus* (Knight): change to *Neolygus omnivagus* (Knight).
- *Lygocoris ostryae* (Knight): change to *Neolygus ostryae* (Knight).
- *Lygocoris parrotti* (Knight): change to *Neolygus parrotti* (Knight).
- *Lygocoris parshleyi* (Knight): change to *Neolygus parshleyi* (Knight).
- *Lygocoris piceicola* (Kelton): change to *Neolygus piceicola* (Kelton).
- *Lygocoris quercalbae* (Knight): change to *Neolygus quercalbae* (Knight).
- *Lygocoris semivittatus* (Knight): change to *Neolygus semivittatus* (Knight).
- *Lygocoris univittatus* (Knight): change to *Neolygus univittatus* (Knight).
- *Lygocoris viburni* (Knight): change to *Neolygus viburni* (Knight).
- *Lygocoris vitticollis* (Reuter): change to *Neolygus vitticollis* (Reuter).
- *Lygocoris walleyi* (Kelton): change to *Neolygus walleyi* (Kelton).

Melanotrichus elongatus Kelton

Orthotylus leonardi was proposed by Kerzhner and Schuh (1995) for *O. elongatus* (Kelton 1980), *M. elongatus* Kelton, a junior secondary homonym of *O. elongatus* Wagner 1965.

It would seem that the name *M. leonardi* (Kerzhner & Schuh) should replace *M. elongatus* Kelton.

Microphylellus adustus binotatus Knight

This species was synonymized with *Reuteroscopus falcatus* Van Duzee by Schuh (2001). However, *R. falcatus* Van Duzee was made the type species of the new genus *Vanduzeephylus* by Schuh and Schwartz (2004). Hence, the name *M. adustus binotatus* Knight should be replaced by *Vanduzeephylus falcatus* (Van Duzee).

Microphylellus flavipes (Provancher)

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it should now be called *Plagiognathus flavipes* (Provancher).

Microphylellus longirostris (Knight)

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it should now be called *Plagiognathus longirostris* (Knight).

Microphylellus maculipennis Knight

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it is now called *Plagiognathus maculipennis* (Knight).

Microphylellus modestus Reuter

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it is now called *Plagiognathus modestus* (Reuter).

Microphylellus tsugae Knight

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, this is now *Plagiognathus tsugae* (Knight).

Microphylellus tumidifrons Knight

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it is now *Plagiognathus tumidifrons* (Knight).

Parapsallus vitellinus (Scholtz)

This introduced species was transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it should be called *Plagiognathus vitellinus* (Scholtz).

Pinophylus rolfsi (Knight)

This is now *P. alpinus* (Van Duzee) according to Schwartz (2013), as noted under *Dacota hesperia* Uhler above. Hence, a nomenclature change is necessary.

Platylygus Van Duzee

Pappus Distant has been shown to be the senior synonym of *Platylygus* Van Duzee by Henry (2006). Thus, all five species of *Platylygus* should be transferred to *Pappus*:

- *Platylygus luridus* (Reuter): change to *Pappus luridus* (Reuter).
- *Platylygus piceicola* Kelton: change to *Pappus piceicola* (Kelton).
- *Platylygus pseudotsugae* Kelton: change to *Pappus pseudotsugae* (Kelton).
- *Platylygus rolfsi* Knight: change to *Pappus rolfsi* (Knight).
- *Platylygus rubripes* Knight: change to *Pappus rubripes* (Knight).

Plesiodema sericeum (Heidemann)

Plesiodema sericeum Heidemann has been placed as the type species of the new genus *Izyaius* by Schwartz (2006). Hence, the name *P. sericeum* should be changed to *Izyaius sericeum* (Heidemann).

Psallus alnicenatus Knight

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it should now be called *Plagiognathus alnicenatus* (Knight).

Psallus morrisoni Knight

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it should now be called *Plagiognathus morrisoni* (Knight).

Psallus parshleyi Knight

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it should now be called *Plagiognathus parshleyi* (Knight).

Psallus physocarpi Henry

This species has been transferred to the genus *Plagiognathus* Fieber by Schuh (2001). Hence, it should now be called *Plagiognathus physocarpi* (Henry).

Sthenarus cuneotinctus Van Duzee

Schuh and Schwartz (2004) have made this species the type of the new genus *Aurantiocoris*. Hence, the species should now be cited as *Aurantiocoris cuneotinctus* (Van Duzee).

Teleorhinus brindleyi Knight

This species was synonymized with *T. cyaneus* Uhler by Wyniger (2010). Hence, the name should be changed to *T. cyaneus* Uhler.

Family TINGIDAE

Dictyonota tricornis (Schrank)

The genus *Kalama* Puton was recognized by Péricart (1982), with *Kalama tricornis* (Schrank) being one of the included species (Froeschner 2001). This latter species was recorded as introduced into Canada and the United States by Drake and Ruhoff (1965) under the name *D.* (*Alcletha*) *tricornis* (Schrank), a fact reiterated by Froeschner (2001). Hence, this tingid should now be recorded as an introduction under the name *K. tricornis* (Schrank).

Family OXYCARENIDAE

Crophius ramosus Barber

Henry *et al.* (2015) resurrected the genus *Mayana* Distant and cited *Mayana ramosa* (Barber) as one of the included species. Hence, *C. ramosus* Barber should be changed to *M. ramosa* (Barber).

Family PIESMATIDAE

Piesma cinereum (Say)

Péricart (1974) made *Tingis cinerea* Say the type species of a new subgenus that he named *Parapiesma*, and *Parapiesma* Péricart was raised to generic status by Heiss and Péricart (1997). Hence, *Parapiesma cinereum* (Say) is the current name for what was previously called *Piesma cinereum* (Say).

Piesma explanatum McAtee

This piesmatid was included in the subgenus *Parapiesma* by Péricart (1974). Since *Parapiesma* Péricart was raised to generic status by Heiss and Péricart (1997), the current name for this taxon, previously called *Piesma explanatum* McAtee, is *Parapiesma explanatum* (McAtee).

Family PENTATOMIDAE

Genus *Acrosternum*, subgenus *Chinavia* Orian

Chinavia Orian was treated as a distinct genus by Ahmad *et al.* (1996). This results in the following changes:

- *A. hilare* (Say): change to *C. hilaris* (Say).
- *A. pensylvanicum* (Gmelin): change to *C. pensylvanica* (Gmelin).

Apateticus bracteatus (Fitch)

Thomas (1992) recognized the genus *Apoecilus* Stål separate from *Apateticus* Dallas and keyed *Apoecilus bracteatus* Fitch. This is the name that should be recognized for this species.

Apateticus cynicus (Say)

This species should now be called *Apoecilus cynicus* (Say), as noted above.

Codophila remota (Horváth)

Kerzhner (1993b) and Rider (1998) have noted that the correct name for this taxon is *Anthemia eurynota remota* (Horváth).

Cosmopepla bimaculata (Thomas)

Rider and Rolston (1995) have noted that the correct name for the species is *C. lintneriana* Kirkaldy.

Holcostethus piceus (Dallas)

Rider and Rolston (1995) proposed the new name *H. macdonaldi* for this species.

III. Spelling Errors

Family CORIXIDAE

Hesperocorixa harrisi (Uhler)

Should be *Hesperocorixa harrisii* (Uhler), according to Jansson (2002).

Hesperocorixa kennicotti (Uhler)

Should be *Hesperocorixa kennicottii* (Uhler), according to Jansson (2002).

Family MIRIDAE

Actinocoris Reuter

This should be spelt *Actitocoris* Reuter.

Atractotomus cerocarpi Knight

This should be spelt *Atractotomus cercocarpi* Knight.

Closterotomus norvegicus (Gmelin)

This should be spelt *Closterotomus norwegicus* (Gmelin), according to Kerzhner and Josifor (1999).

Family TINGIDAE

Alveotingis grossocerata Osborne & Drake

Should be *Alveotingis grossocerata* Osborn & Drake.

Family LYGAEIDAE

Melanopleurus pyrropterus (Stål)

This should be spelt *Melanopleurus pyrrhopterus* (Stål).

IV. Other Comments

Family ANTHOCORIDAE

Anthocoris tomentosus Péricart

Lewis and Horton (2012) have shown that many of the occurrence records listed from *A. tomentosus* from the Yukon by Scudder (1997) are a new species that was described as *A. aquilivenis* Lewis. Lewis and Horton (2012) also gave records of *A. aquilivenis* for Alaska and British Columbia that had previously been determined as *A. tomentosus*. However, *A. tomentosus* still has valid occurrence records from Alaska, Yukon and British Columbia.

Orius diespeter Herring

The Yukon records for *O. diespeter* Herring given in Scudder (1997) are in fact the species *O. sibericus* Wagner (Lewis *et al.* 2015). Hence, the Yukon record for *O. diespeter* in Scudder (1997) should be deleted and replaced by *O. sibericus*. However, *O. diespeter* Herring does occur in the Yukon (Lewis and Horton 2010), although it is recorded as *O. tristicolor* (White) by Scudder (1997) (see below).

Orius tristicolor (White)

Lewis and Horton (2010) have shown that all records from the Yukon listed by Scudder (1997) as *O. tristicolor* are in fact a colour variation of *O. diespeter* Herring. Lewis and Horton (2010) also suggest that all records of *O. tristicolor* in eastern Canada

actually refer to *O. diespeter*. Hence, *O. tristicolor* is deleted for Saskatchewan to Newfoundland, and replaced by *O. diespeter*.

Lewis and Horton (2010) updated the known distribution of *O. diespeter* to include Alberta, British Columbia, Nova Scotia, Ontario, Quebec, the Yukon, and Alaska: *O. tristicolor* was recorded from Alberta and British Columbia but not Alaska.

Lewis informed me on January 30, 2018 (in litt.) that a male specimen in the UBCZ collection, number ANTH0784, with data "Firth R., 69°08'N 140°14'W, 23.vi.1984 (S.G. Cannings)" that she determined in 2010 is in fact *O. tristicolor*. This was originally reported in Lewis and Horton (2010) as *O. diespeter*. However, this has been clarified since by Lewis (in litt., 23 February 2018). Hence, *O. tristicolor* is still recorded from the Yukon but not Alaska.

Family MIRIDAE

Tupiocoris agilis (Uhler)

Tupiocoris agilis was first reported from British Columbia by Parshley (1919) as *Dicyphus agilis* Uhler with records for Saanich Dist., V.I., Apr. 30, Sept. 14, 1918 (W. Downes). It was also reported from British Columbia by Downes (1927) under the same name, with specimens recorded from Agassiz, Sept. 1921 (R. Glendenning), Duncan, Aug. 4th, 1921 (W. Downes) and Saanich, June 18th, 1918 (W. Downes). Kelton (1980) writing under *D. confusus* Kelton, concluded that the early records of what is now *T. agilis* (Uhler) probably refer to what is now *T. confusus* (Kelton), *T. similis* (Kelton), or some other species.

However, new records of *T. agilis* (Uhler) for British Columbia were published by Schwartz and Scudder (2001). Although I have been unable to trace the earlier specimen listed by Parshley (1919) and Downes (1927), these can be ignored, as the recent records by Schwartz and Scudder (2001) validate the species in British Columbia.

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The bees of British Columbia (Hymenoptera: Apoidea, Apiformes)

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ABSTRACT

British Columbia is the most biologically diverse province in Canada, and its wide range of landscapes – particularly the dry valley bottoms and basins of the Columbia, Kootenay, Okanagan, Kettle, and Similkameen River systems – make it ideal for many groups of Hymenoptera, including bees. With the exceptions of some generic- or family-level treatments, no comprehensive account of the bees of British Columbia has been published, although recent studies have indicated that more than half of Canada's bee species may be found in the province, with many of these found nowhere else in the country.

Here, we summarize the province's bee fauna by providing a comprehensive annotated checklist of species. For each species, we indicate the ecozone(s) in which they are presently known to occur, and we provide summary statistics and analyses to compare ecozones. We also summarize the growth in knowledge of the province's bee species over time, and all species accounts for the province are accompanied by a list of supporting literature or data. Although we feel this list is comprehensive, it is likely that we have overlooked some published accounts, and additional undocumented species will show up.

In total, we record 483 bee species from British Columbia, 37 of which are considered new to the province. Among these, 20 species (or subspecies) are recorded as new to Canada, including: *Andrena* (*Euandrena*) *misella* Timberlake, *Panurginus cressoniellus* Cockerell [Andrenidae], *Lasioglossum* (*Dialictus*) *obnubilum* (Sandhouse), *L.* (*Evylaeus*) *argemonis* (Cockerell), *L.* (*Hemihalictus*) *glabriventre* (Crawford), *L.* (*Hemihalictus*) *kincaidii* (Cockerell) [Halictidae], *Osmia* (*Melanosmia*) *laeta* Sandhouse, *O.* (*Melanosmia*) *malina* Cockerell, *O.* (*Melanosmia*) *pulsatillae* Cockerell, *O.* (*Melanosmia*) *raritatis* Michener, *Anthidium* (*Anthidium*) *formosum* Cresson, *Dianthidium* (*Dianthidium*) *plenum plenum* Timberlake, *D.* (*Dianthidium*) *singulare* (Cresson), *Stelis* (*Stelis*) *ashmeadiellae* Timberlake, *S.* (*Stelis*) *calliphorina* (Cockerell), *Dioxys pomonae pomonae* Cockerell, *Megachile pugnata pomonae* Cockerell [Megachilidae], *Nomada crotchii* Cresson, *Melissodes* (*Eumelissodes*) *saponellus* Cockerell, and *Habropoda miserabilis* (Cresson) [Apidae].

Key words: Andrenidae, Apidae, Colletidae, Halictidae, Megachilidae, Melittidae, diversity

INTRODUCTION

British Columbia is a vast landscape with variable topography, geology, and climate that enable the largest total biodiversity of any province or territory in the country (Cannings and Cannings 2015; Canadian Endangered Species Conservation Council 2016). Approximately 80,000 species are estimated to live in Canada (Canadian Endangered Species Conservation Council 2016), with more than 50,000 species occurring in British Columbia alone (Cannings and Cannings 2015). However, precise knowledge comes only from fully documenting the species that have been recorded via faunal checklists. In addition to providing data for increasing faunistic knowledge, species checklists provide important baselines for assigning a species' conservation status and enabling the prioritization of habitat protection, management, conservation and land

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use decisions. For many invertebrate groups, species checklists do not exist or are incomplete, although the completion of *Wild Species 2015* (Canadian Endangered Species Conservation Council 2016) has enabled a better understanding of the provincial and territorial diversity across the country for many taxa, including in British Columbia.

In the last decade pollinators, particularly bees, have come to the forefront of conservation importance due to their integral link to pollination, food supply and overall ecosystem health. A key component to assessing the conservation status of bee communities begins with understanding the species present, their respective range extents, and potential habitat associations according to the ecosystem mapping throughout the species' range. The range extents for many bee species recorded from British Columbia are unclear, and more inventories are needed to better define their limits (Heron and Sheffield 2015). The inventory for bee species in British Columbia is incomplete, and most past efforts to compile species lists have focused on documenting a narrow range of taxa (e.g., Buckell 1949, 1950, 1951; Cannings 2011 - *Bombus*), or have not been comprehensive (e.g., Viereck *et al.* 1904a–d, 1905a, b, 1906). More recently, studies providing species information have been ecological in nature and have focussed within a limited geography (e.g., Elwell *et al.* 2016). However, bee diversity estimates for British Columbia have been treated in a more general sense: the province is known to have the highest bee diversity in Canada, with estimates ranging from 369 (Sheffield *et al.* 2014) to possibly more than 600 species (Sheffield *et al.* 2017), the latter estimate being based on DNA barcoding results.

Numerous factors likely contribute to this high biodiversity. For example, bees are closely associated with plant diversity and habitat type. Approximately 2,500 native vascular plants have been recorded from British Columbia (Douglas *et al.* 2002; British Columbia Conservation Data Centre 2018), some of which are part of rare ecosystems and plant communities unique to Canada (Straley *et al.* 1985). The southern part of the province is also the northernmost extension of numerous unique southern ecosystems, allowing numerous bee species to range into these same areas. Many of these bee species are solitary and depend on specific soil and climate variables that define or seemingly restrict their range (Sheffield *et al.* 2014); the Western Interior Basin for example, though by far the smallest ecozone in Canada, contains a significant number of the country's bee species, some of which occur nowhere else in Canada (Sheffield *et al.* 2014). Though no comprehensive checklist of British Columbia bee species has been previously completed (although see Sheffield and Heron 2017), some components of the province's bee fauna were covered, as indicated above. In addition, Tepedino and Griswold (1995) provided a list of species for the Columbia Basin, which included some specimens from British Columbia.

Our objective here is to provide the first published, comprehensive list of the bees of British Columbia, correcting, updating, and validating occurrence data in lists previously provided to the Canadian Endangered Species Conservation Council (2016) and E-Fauna (Sheffield and Heron 2017). Species occurences in the province are fully documented with references to literature, and links to datasets are provided. This project also contributes to the overall knowledge of apoid wasps in the province; the Spheciformes treated recently by Ratzlaff (2015) and Ratzlaff *et al.* (2016) and all studies building on the provincial summary of Apoidea provided by Cannings and Scudder (2001).

MATERIALS AND METHODS

Most of the data presented here were compiled from published literature, ranging from published taxonomic treatments, species lists, ecological studies, and unpublished graduate theses. In addition, data were also mined from websites and non-peer-reviewed or unpublished studies (i.e., grey literature) and verified with specimen or photographic evidence. Our list builds on previous faunistic work that has focused on northwestern North American, including British Columbia (Viereck *et al.* 1904a–d, 1905a, b; 1906),

and later works specific to the province (Buckell 1949, 1950, 1951), much of which was compiled for the *Wild Species 2015* national assessment (Canadian Endangered Species Conservation Council 2016). In cases where records for "BC" were recorded in the literature (e.g., Hurd 1979) without accompanying data, the "species x British Columbia" were entered as search terms in Biodiversity Heritage Library (https://www.biodiversitylibrary.org/); however, in a few instances, no supporting literature/data could be found. References and notes supporting the presence of each species in the province are given next to each taxon in Supplemental Material.

Data were also compiled from many past and more recent collection efforts in the province, including studies being conducted by the British Columbia Ministry of Environment and Climate Change Strategy (JMH), Royal Saskatchewan Museum (CSS), and past studies conducted out of York University (Toronto, ON). Much of this recent material was used in the Barcodes of Life campaign for the bees of Canada (Sheffield *et al.* 2017). In addition, many specimens were examined from the Royal British Columbia Museum (Victoria, BC), the Spencer Entomology Museum, University of British Columbia (Vancouver, BC), the Royal Saskatchewan Museum (Regina, SK), York University (Toronto, ON), and the Canadian National Collection of Insects, Arachnids, and Nematodes (Ottawa, ON). The complete species list has been added to Canadensys (http://www.canadensys.net/) at https://doi.org/10.5886/NKZFXC, and has been registered with GBIF [assigned the following GBIF UUID: 7b944cc6-1ffa-49de-aab8-2a5ab543422b]. Occurrence data from species recorded as new to the province and/or country have also been added to Canadensys [https://doi.org/10.5886/INGA8Z] and is also registered with GBIF [GBIF UUID: f9c49aed-ba4b-454e-b88a-cbe1fff5b2b6]. An updated version of the list will also be maintained at the Bees of Canada website: http://www.beesofcanada.com/home.

Although some of the literature sources examined (e.g., Mitchell 1960, 1962; Hurd 1979; Cannings 2011) list a species as only occurring in the province, we specifically tried to mine data that would provide geographic information to allow us to assign each species to the Canadian ecozones represented in the province (see Ecological Stratification Working Group 1995; Environment and Climate Change Canada 2016). Canada's terrestrial land base is classified into 15 ecozones that are part of a broad ecological framework for North America (Ecological Stratification Working Group 1995; Wilken *et al.* 1996; Commission for Environmental Cooperation 1997) that classify a geographic area of the country with similar physiography, hydrology, climate, wildlife potential and vegetation. The attributes of each ecozone promote classification based on unique assemblages of plant and animal communites based on climate zones and soils. The ecozones in which each bee species occurs provides additional ecological information that may provide conservation value. The six Canadian ecozones represented in British Columbia are the Pacific Maritime [PacM], Western Interior Basin [WIB], Montane Cordillera [MonC], Boreal Plains [BorPl], Boreal Cordillera [BorC], and Taiga Plains [TaiPl]. Information on each ecozone in British Columbia is summarized from the references above.

The Pacific Maritime [PacM] ecozone has an area of 195,000 km^2, and occurs along the west coast (including coastal islands) from the United States (Washington) border in the south, northwards to the Alaska Panhandle. This ecozone is the wettest in the Canada, with extensive areas of temperate old-growth coniferous forests (i.e., western redcedar (*Thuja plicata* Donn ex D. Don), yellow-cedar (*Cupressus nootkatensis* D. Don), western hemlock (*Tsuga heterophyla* (Raf.) Sarg.), mountain hemlock (*Tsuga mertensiana* (Bong.) Carr.), Douglas-fir (*Pseudotsuga menziesii* (Mirb.) Franco), Pacific silver fir (*Abies amabilis* Douglas ex J. Forbes), and Sitka spruce (*Picea sitchensis* (Bong.) Carr.), with high mountains with alpine tundra and glacial, and lowland estuary and valley-bottom floodplain habitats (Fig. 1). It contains numerous rare and endangered ecosystems, including Garry Oak (*Quercus garryana* Douglas ex Hook.) and associated

ecosystems, sparsely vegetated coastal sand ecosystems, bog and wetland habitats, and the lowland riparian forests of the Fraser Valley.

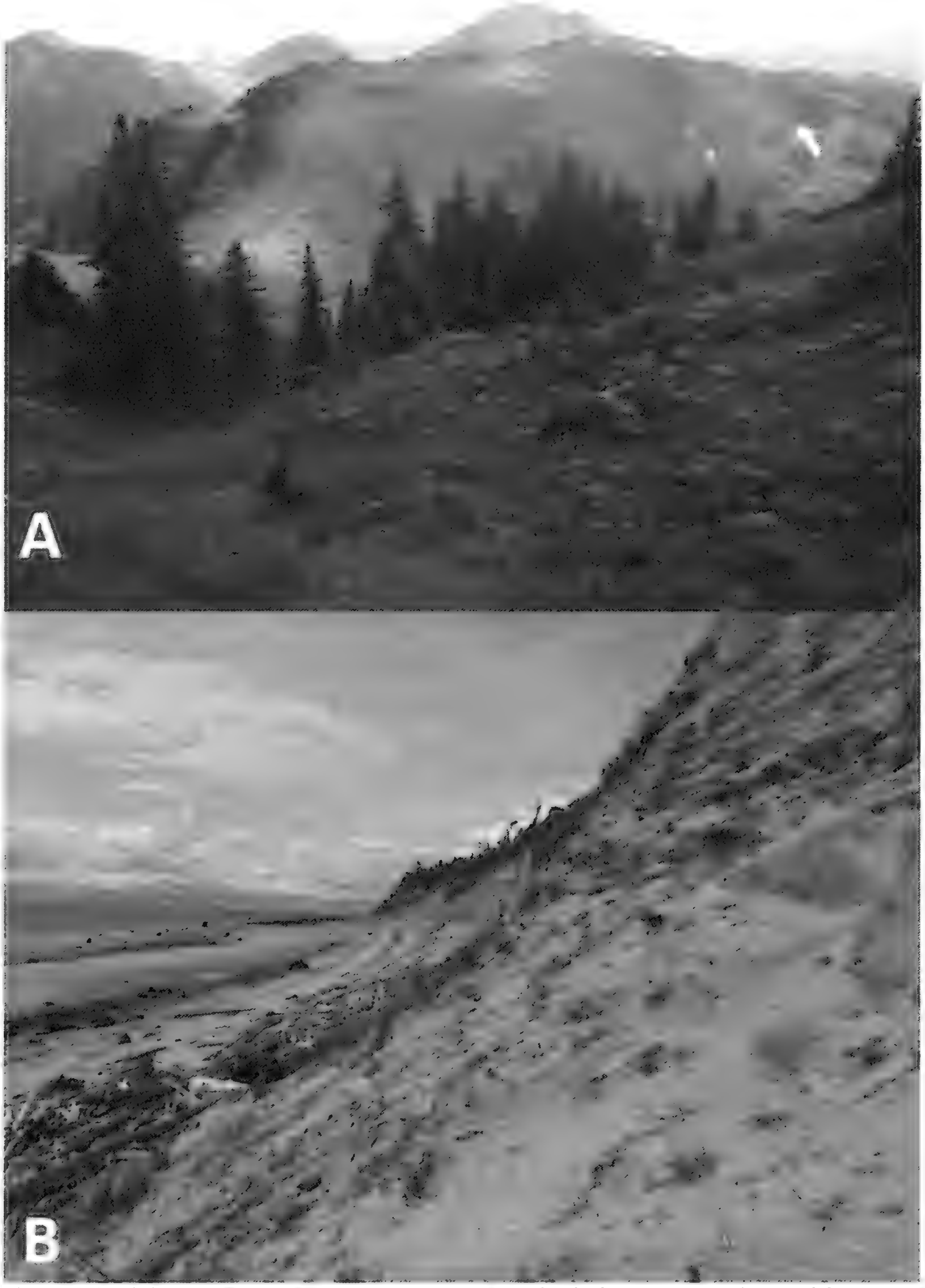

Figure 1. Pacific Maritime [PacM] ecozone. A) subalpine coastal coniferous forests, Greig Ridge, Strathcona Provincial Park. Photo J. Heron; B) coastal sand ecosystem on south side of Savary Island. Photo J. Heron.

The Western Interior Basin [WIB, also called the Semi-Arid Plateau] is the smallest ecozone in Canada (previously classification considered this ecozone part of the Montane Cordillera), and all 56,466 km^2 are restricted to the south–central part of the province. The boundary of this ecozone is comparable to the Southern Interior Ecoprovince of the province's Ecoregion Classification System. The ecozone (Fig. 2) represents the northernmost extension of the Great Basin Sagebrush Desert Biome that stretches from British Columbia through the Midwestern United States to Mexico. Approximately 2% of the land area of this ecozone is classified as native grasslands and 73% as forests. There are a number of species at risk that are confined to the WIB and, more specifically, to the low-elevation plant communities of this ecozone. The cumulative effects from multiple threats, such as natural habitat conversion, fragmentation, recreational use and invasive species, have led to these species being at risk. In particular, the antelope-brush

(*Purschia tridentata* (Pursh) DC.) plant communities in the south Okanagan Valley have significantly declined in quality and spatial area since the 1800s (Schluter *et al.* 1995; Lea 2001, 2008; Iverson and Haney 2012; Iverson 2012). More specifically, the antelope-brush/needle-and-thread Grass plant community has declined from 9,863 ha in 1800 to 3,217 ha in 2009, a loss of 67.4% of the original extent of this ecosystem (Iverson 2012). More broadly across the WIB, approximately 16% of grasslands (1188km^2) have been converted to urban and agricultural development since 1850 (Wikeem and Wikeem 2004; Grasslands Conservation Council of British Columbia 2004; B.C. Ministry of Environment 2007). Habitat loss continues with high development pressure on undesignated provincial Crown land and natural areas into housing, commercial and agricultural use. Livestock overgrazing is also a threat within provincial Crown lands – both grassland and forested areas.

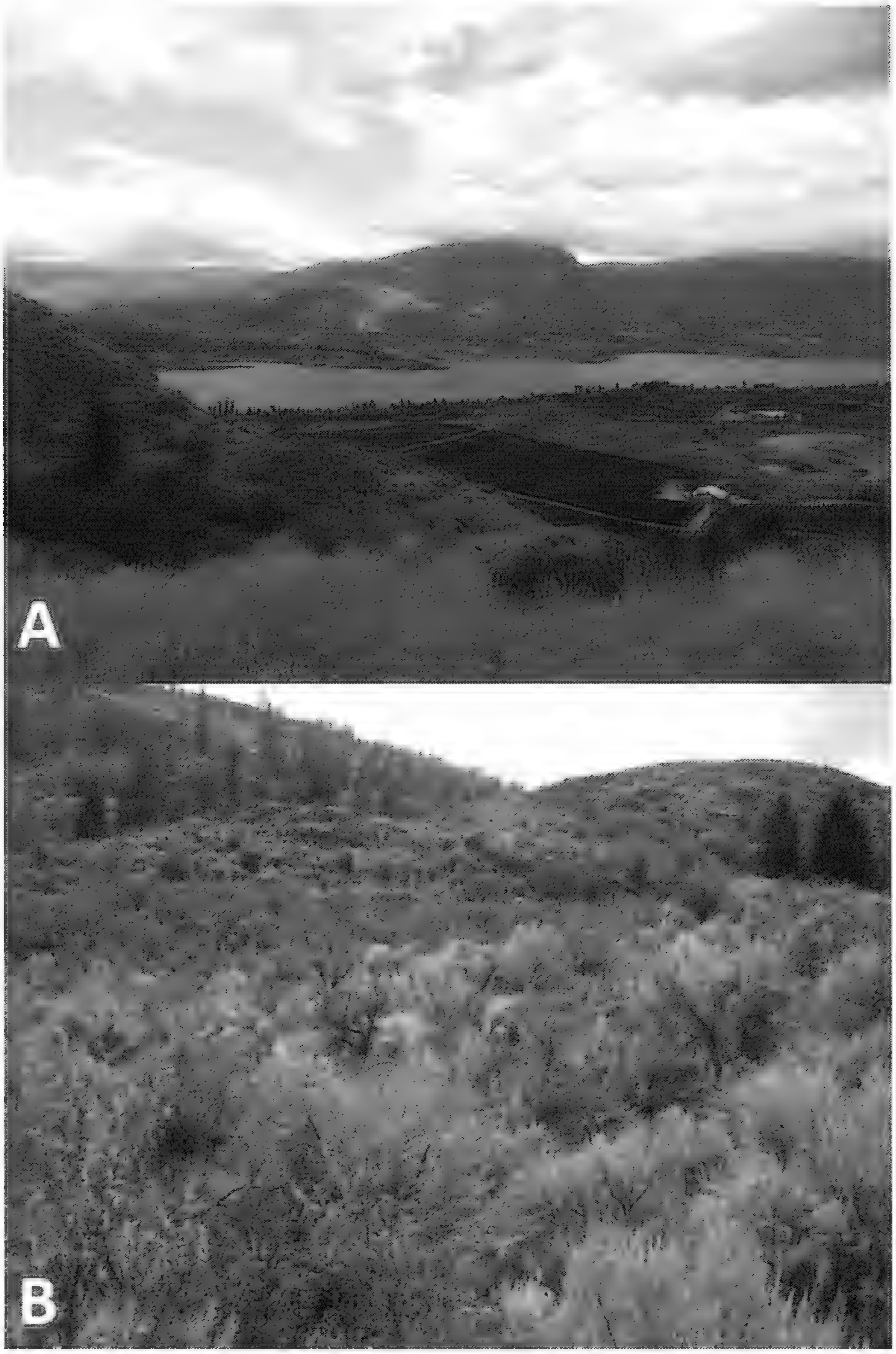

Figure 2. Western Interior Basin [WIB] ecozone. A) lower Okanagan Valley. Photo C.S. Sheffield; B) antelope-brush plant community at Osoyoos Desert Centre, west of Osoyoos. Photo J. Heron.

The British Columbia portion of the Montane Cordillera [MonC] ecozone – about 90% of the total for Canada (Scudder and Smith 2011) – comprises 389,000 km^2. It covers the eastern portion of the province and spans the Rocky Mountains into western Alberta. The ecozone ranges from the United States border to the Skeena Mountains in north–central British Columbia, and includes a broad range of ecosystems, from dense conifer forests to alpine tundra, grasslands, and rugged mountains (Fig. 3): it is likely the most complex ecozone in the province (Scudder and Smith 2011). Approximately 70% of the area is forested, 27% is non-forested, and 3% is water (Scudder and Smith 2011). The climate is characterized by wet winters and dry summers, with mild climate overall throughout the year. The Kootenay region of the province includes the western slopes of the Rocky Mountains, small portions of arid sagebrush and grasslands, fir and cedar forests, large rivers, and numerous valleys that extend southwards into the United States and bring a number of species to their northernmost limits.

Figure 3. Montane Cordillera [MonC] ecozone. A) view south from Cristina Creek. Photo J. Heron; B) Flathead Valley, east of Fernie. Photo J. Heron.

Approximately 5% of the Boreal Plains [BorPl] ecozone occurs in the province (37,940 km^2) and exists across a small portion of north-eastern British Columbia (Fig. 4). More than half of this ecozone (60%) comprises forests and, in British Columbia, the ecozone's other habitats are shrublands and wetlands, as well as native grasslands that have been converted to agricultural areas. Forests grow slowly in the Boreal Plains due to low-nutrient and poorly drained soils and discontinuous permafrost (ESTR Secretariat 2014).

Figure 4. The Boreal Plains [BorPl] ecozone. A) along the Peace River west of Fort St. John enar Hudson's Hope. Photo J. Heron; B) at Pink Mountain, looking northwest at the Rocky Mountains. Photo S. Cannings.

The portion of the Boreal Cordillera [BorC] in British Columbia spans a large portion of the northern half of the province, and stretches into the Yukon. The BorC ecozone (Fig. 5) covers 189,000 km^2 and is dominated by forests of black spruce [*Picea mariana* (Mill.) Britton, Sterns & Poggenburg] and white spruce [*P. glauca* (Moench) Voss], lodgepole pine (*Pinus contorta* Douglas), trembling aspen (*Populus tremuloides* Michx.), balsam poplar (*P. balsamifera* L.) and white birch (*Betula papyrifera* Marshall), with higher-elevation areas of subalpine-fir [*Abies lasiocarpa* (Hooker) Nuttall].

Less than 10% of the Taiga Plains [TaiPl] ecozone occurs in British Columbia – approximately 70,000 km^2. Much of this ecozone (Fig. 6) is boreal spruce forest (68%), wetland, and peatland habitats, with extensive shrub cover (20%). The ecozone also contains some elements of subarctic habitats (ESTR Secretariat 2013).

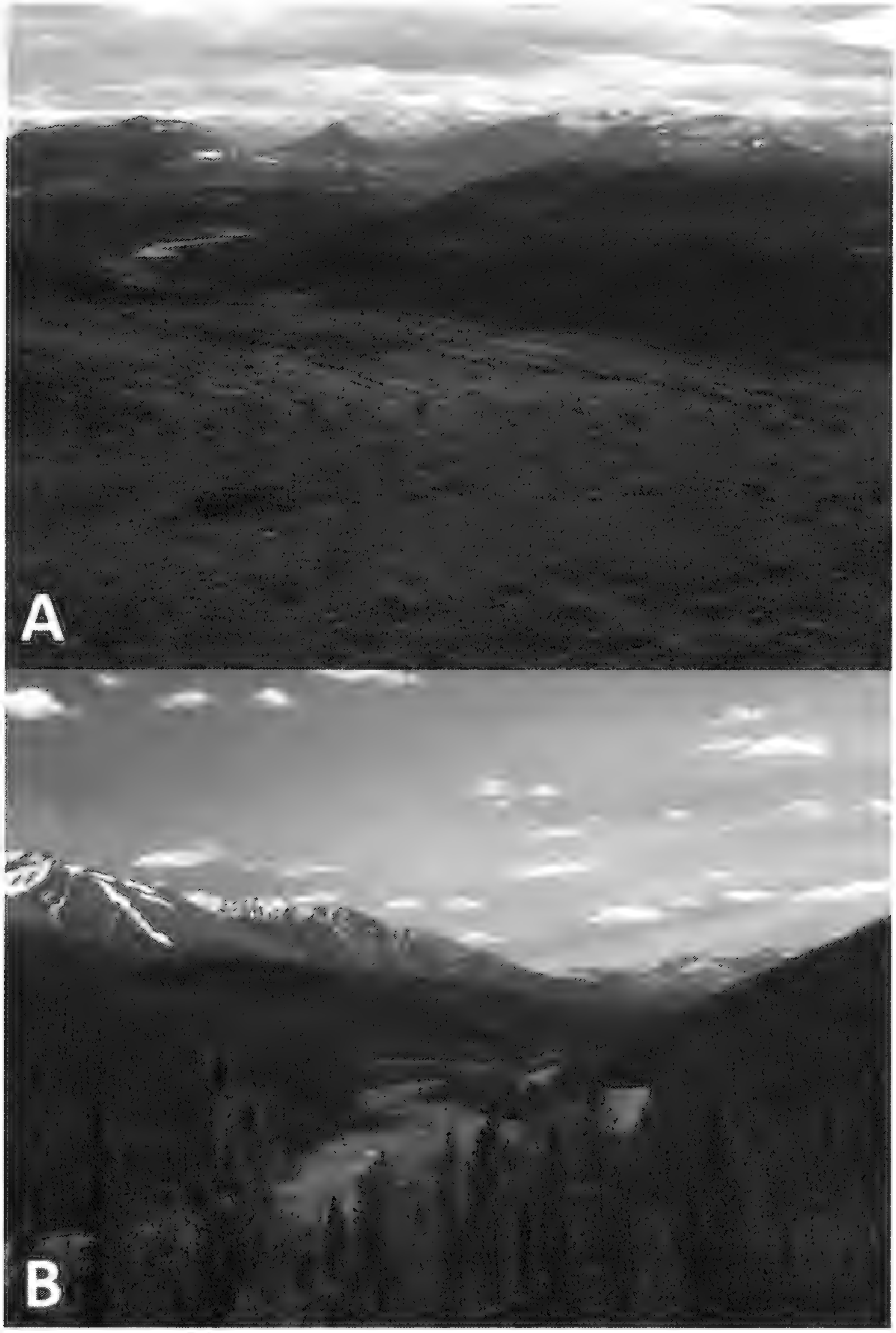

Figure 5. Boreal Cordillera [BorC] ecozone. A) alpine country east of Atlin. Photo S. Cannings; B) North Tetsa River, Stone Mountain Provincial Park. Photo S. Cannings.

The bee fauna of the ecozones in British Columbia were compared both by tallying the species known to occur in each and based on the number of species per 1000 km^2; this latter calculation was done to highlight the diversity of bee species based on the size of each ecozone specifically to draw attention to bee biodiversity hot spots and areas of high conservation value. In addition, a presence/absence matrix of bee species by ecozone was created, and a single link cluster analysis of incidence-based similarity (i.e., Jaccard's index) was performed using Biodiversity Pro (McAleece *et al.* 1997) to explore faunistic similarity of the ecozones occurring in the province.

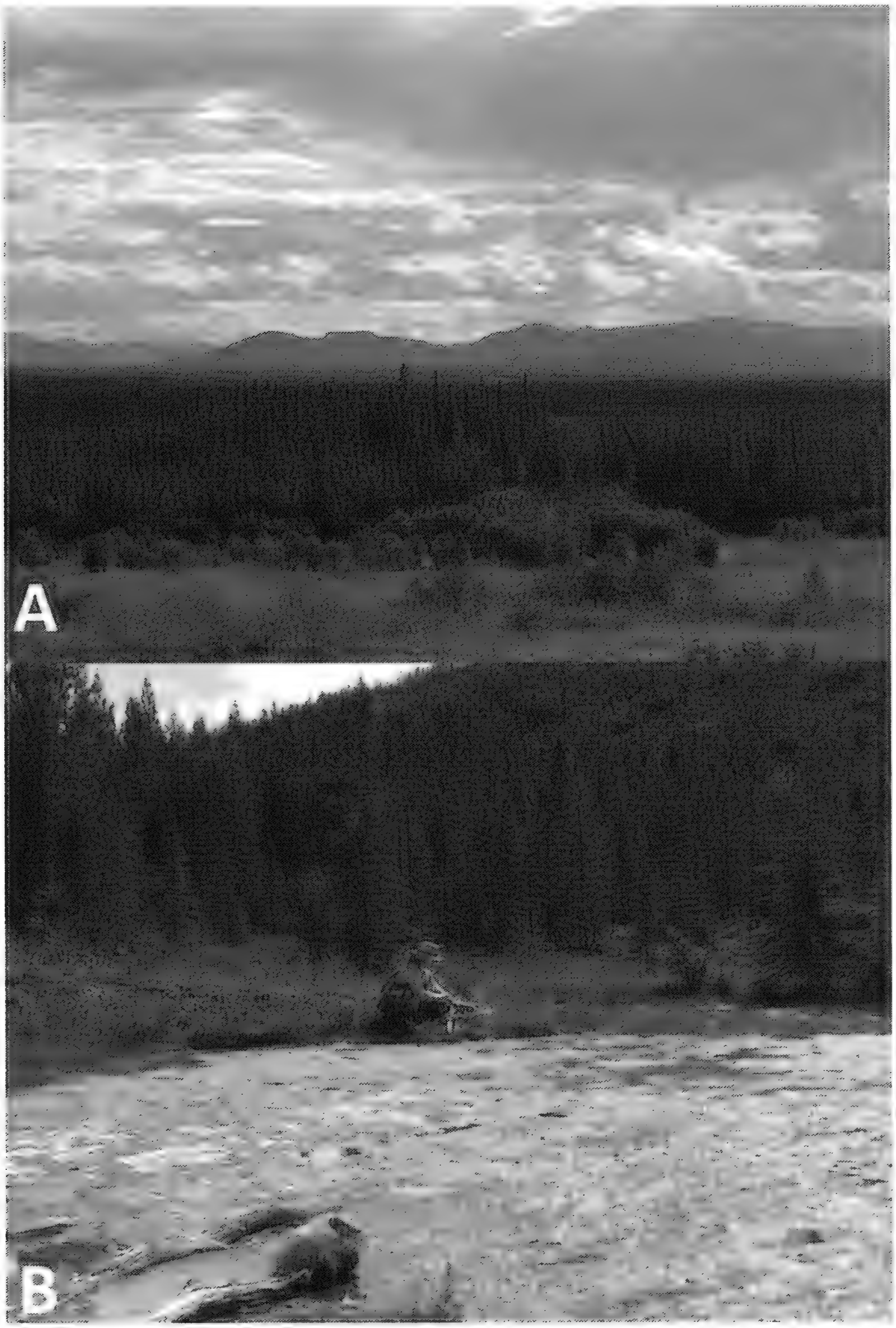

Figure 6. Taiga Plains [TaiPl] ecozone. A) at Fort Nelson, looking west to the Rocky Mountains. Photo S. Cannings; B) Grayling River Hot Spring. Photo C.S. Sheffield.

RESULTS AND DISCUSSION

The first published record of a bee in British Columbia was that of Smith (1861), who described the cuckoo bumble bee, *Apathus* (=*Bombus*) *insularis* (Smith), from the province. Knowledge of the bee fauna of British Columbia has increased dramatically over the last ca. 160 years, with several major published contributions adding greatly to the list of species recorded for the province throughout this period (Fig. 7). The most significant years of contributions (i.e., additions of 20 or more species per year resulting from a single published study or series of related published studies) occurred in the early 1900s (Viereck *et al.* 1904a-d, 1905a, b, 1906; see “A” on Fig. 7), 1924 (Criddle *et al.* 1924; “B” on Figure 7), and 1925 (Sandhouse 1925a, b; “C” on Fig. 7). Yearly increases did not exceed 20 species again until 2010 (Gibbs 2010; “D” on Fig. 7). More recently, Elwell *et al.* (2016) added another 20 species to the provincial list (“E” on Fig. 7). In the present study, we add an additional 37 species (“F” on Fig. 7), 20 of which are new for Canada, for a cumulative provincial count of 483 bee species (Fig. 7). The Megachilidae is the family most well represented, with more than 150 species found in the province,

followed by Andrenidae (largely the genus *Andrena* Fabricius) and Apidae, both with more than 100 species, and Halictidae (Fig. 8).

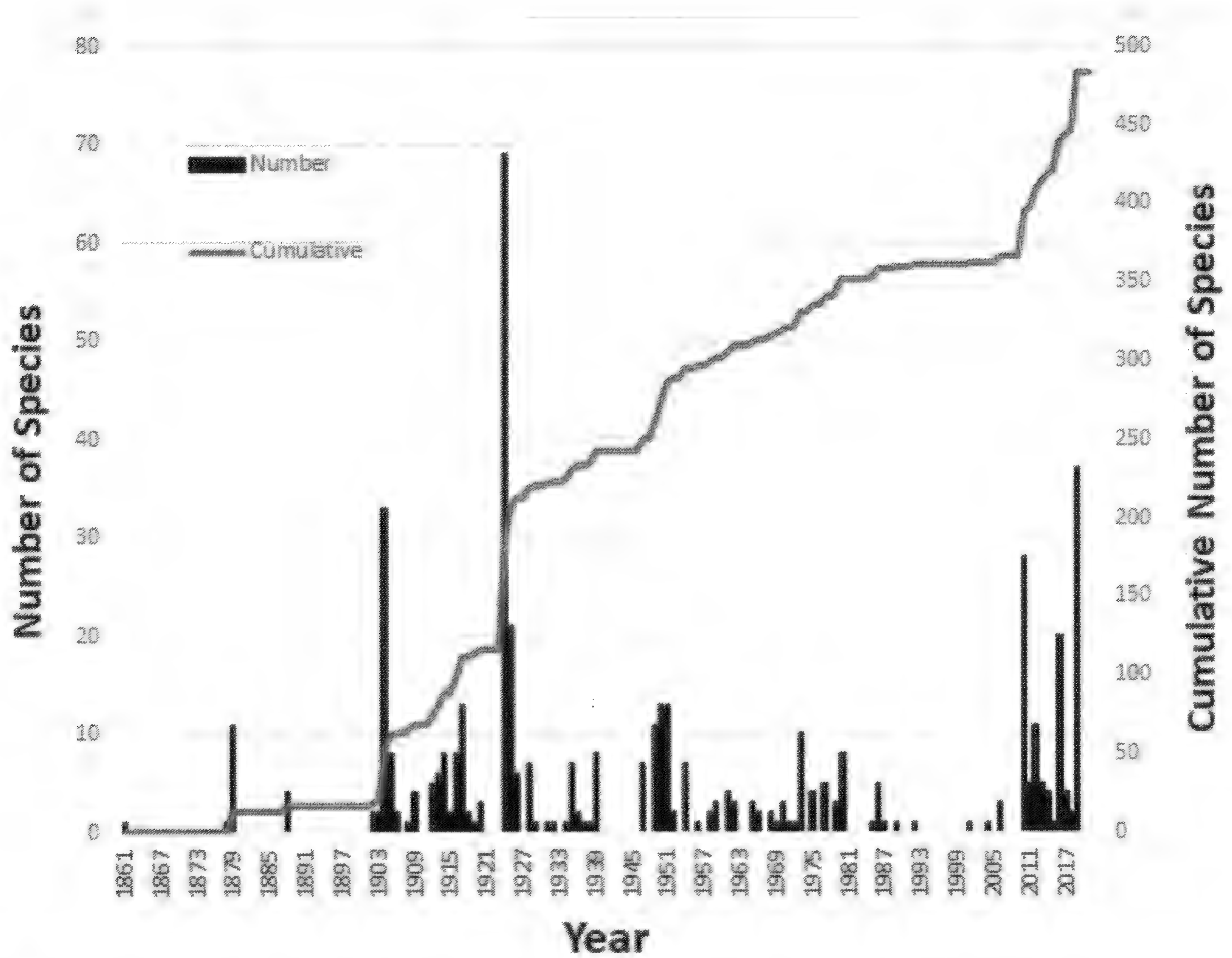

Figure 7. The year by year addition and cumulative total of bee species in British Columbia based on published literature records and other data from 1861 to present (see links to data sets above). Black bars show the number of new species records for each year (i.e., based on the earliest recorded occurrence in the province, see Supplemental Material) (left axis); red line shows the cumulative number of species (right axis) based on these additions. Letters adjacent to bars represent published studies where 20 or more species were added as the result of one publication or group of related publications. A=Viereck et al. 1904a-d (32 species) + Vachal 1904 (6 species); B=Criddle et al. 1924 (56 species) + Sandhouse 1924 (4 species) + Viereck 1924 (11 species); C=Sandhouse 1925a-b (20 species); D=Gibbs 2010 (27 species) + Rightmyer 2010 (1 species); E=Elwell et al. 2016 (20 species); F=present study (37 species).

The rapid growth in species numbers observed in the past 10 years has largely been facilitated through surveys (Heron and Sheffield 2015; Elwell *et al.* 2016), taxonomic revisions (Gibbs 2010; Sheffield *et al.* 2011), and DNA barcoding (Sheffield *et al.* 2017), with a large proportion of species occurring in British Columbia having sequences in the Barcodes of Life Data system (BOLD) with specimens contributed by the Royal Saskatchewan Museum, York University, Simon Fraser University, and the Royal British Columbia Museum, and collecting efforts of the authors and associated researchers at these institutions. These efforts have verified many previous records of others (see Supplemental Material) and have added new records to the province (Gibbs 2010; Heron and Sheffield 2015; Elwell *et al.* 2016). The DNA barcoding efforts have also highlighted the fact that there is still much taxonomic work to do with the British Columbia bee fauna, especially with the cleptoparasitic genera *Sphecodes* (Halictidae) and *Nomada* (Apidae) (Sheffield *et al.* 2017). A recent estimate (Sheffield *et al.* 2017) suggests there could be upwards of 600 species in the province – almost three-quarters of the total for

Canada – with the vast majority of these found in the WIB ecozone (Fig. 9). This is supported by previous estimates of bee diversity in the Columbia Basin in the adjacent USA, which suggests almost 650 species (Mayer *et al.* 2000; Niwa *et al.* 2001), with estimates as high as 1,000 species (Tepedino and Griswold 1995).

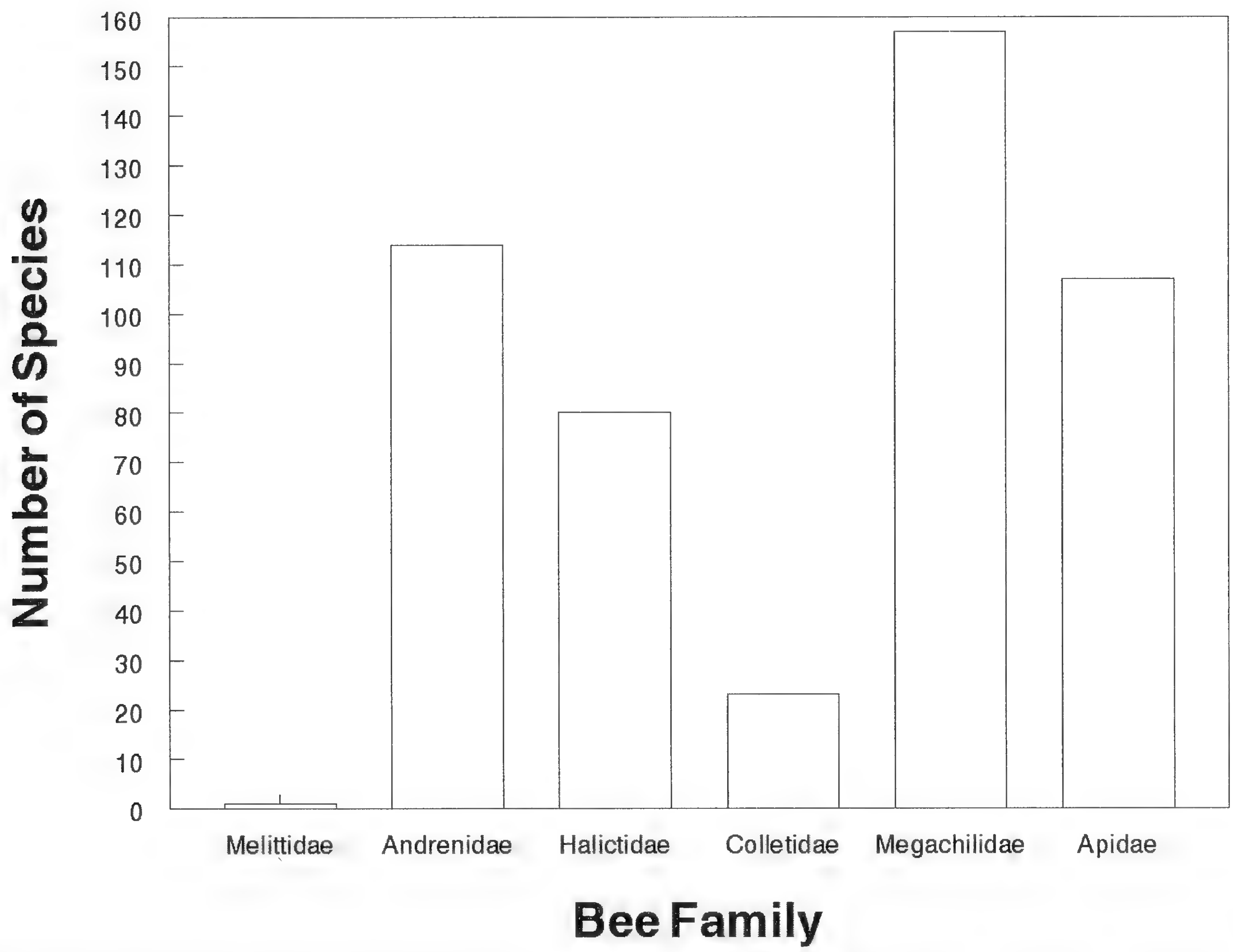

Figure 8. The number of species currently recorded for each bee family in British Columbia.

Although not as diverse as some other North American bee hot spots (Carril *et al.* 2018), the WIB ecozone is the most diverse for bees, with 411 confirmed species (Fig. 9) – almost half of those known from Canada – with 176 of these not occurring in the province's other ecozones (and most of these species are not found anywhere else in Canada). The PacM and MonC ecozones are also diverse with respect to bees, with 207 and 204 confirmed species in each, respectively (Fig. 9). The PacM ecozone has 26 bee species not yet reported elsewhere in the province, with an additional 18 seemingly restricted to the MonC ecozone within the province. These southern ecozones (i.e., PacM, WIB, MonC) have higher levels of similarity to each other than to more northerly ecozones (i.e., TaiPl, BorC, BorPl; Fig. 10), although the bee fauna of the WIB shared less than 37% of its species with the MonC and PacM. This low level of similarity is due to the large number of species endemic to the WIB within Canada, supporting the suggestion that this small area has very high conservation value (South Okanagan Similkameen Conservation Program 2012), especially for bees in Canada (Fig. 9). The British Columbian segments of the three other ecozones are much less speciose, with no bee species seemingly restricted to any one specific ecozone; all three ecozones share more than 50% of their species (Fig. 10). The BorPl ecozone has 88 recorded bee species, the BorC has 73 bee species, and the TaiPl contains 70 bee species. The northern *Bombus occidentalis mckayi* Ashmead, with a national conservation status by the Committee on the Status of Endangered Wildlife in Canada (COSEWIC) of Special

Concern in Canada (COSEWIC 2014; Sheffield *et al.* 2016), is seemingly restricted to the BorC ecozone within the province.

For the overall checklist structure below, we follow Sann *et al.* (2018) for family placement and, for convenience, we follow Michener (2007) for within-family classification, except for the genus *Lasioglossum* Curtis, which follows Gibbs *et al.* (2013). New records for the province are indicated with an "*", new records for Canada are indicated with an "†". These specimens are usually supported by material in the Barcodes of Life Data (BOLD) System (and see Sheffield *et al.* 2017), although notes are also provided in the supplementary links provided above. Species notes and other annotations are provided for some species to clarify their status in the province.

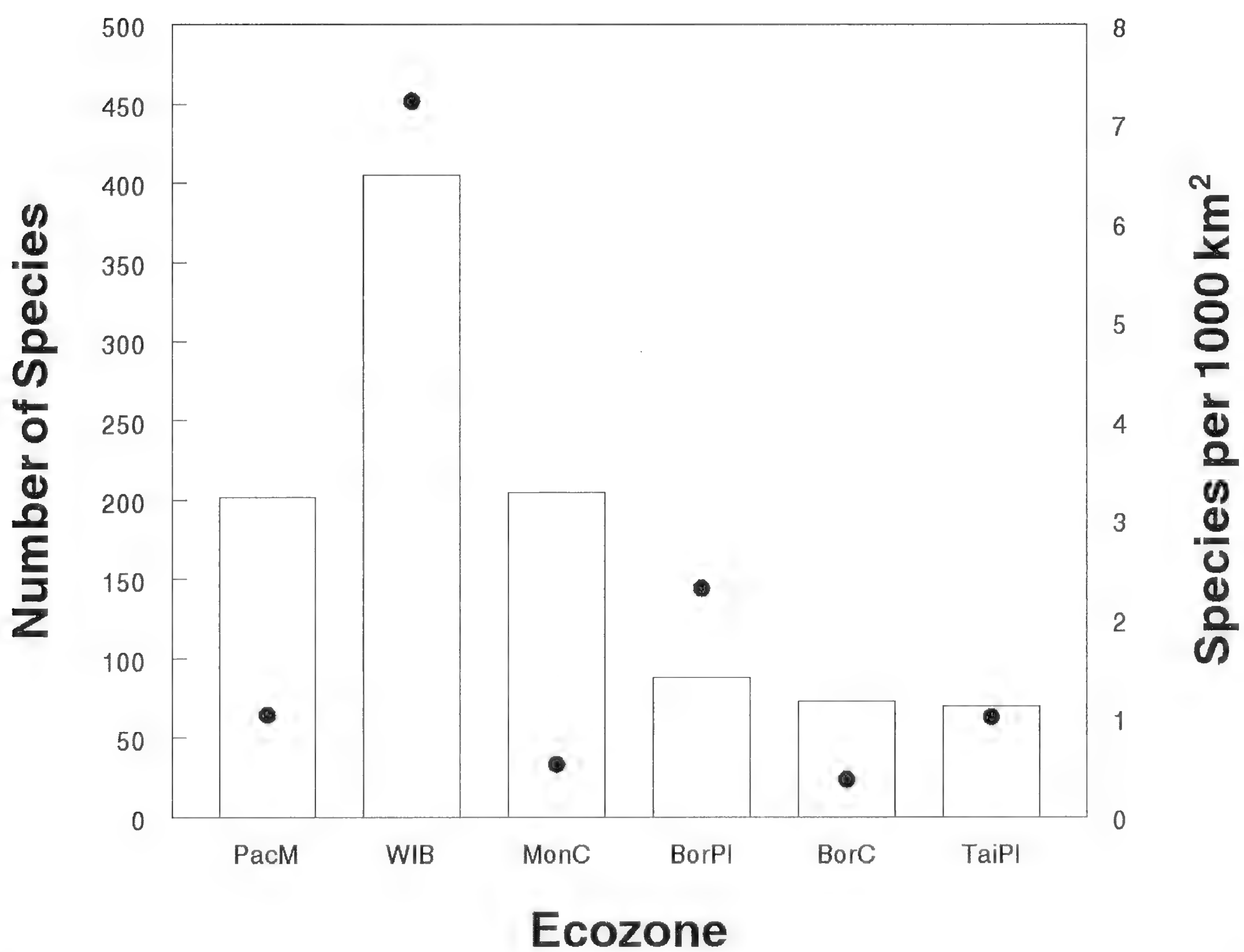

Figure 9. The number of species (bars, left Y-axis) and species/1000km^2 (dots, right Y-axis) for each ecozone in British Columbia. PacM=Pacific Maritime; WIB=Western Interior Basin; MonC=Montane Cordillera; BorPl=Boreal Plains; BorC=Boreal Cordillera; TaiPl=Taiga Plains.

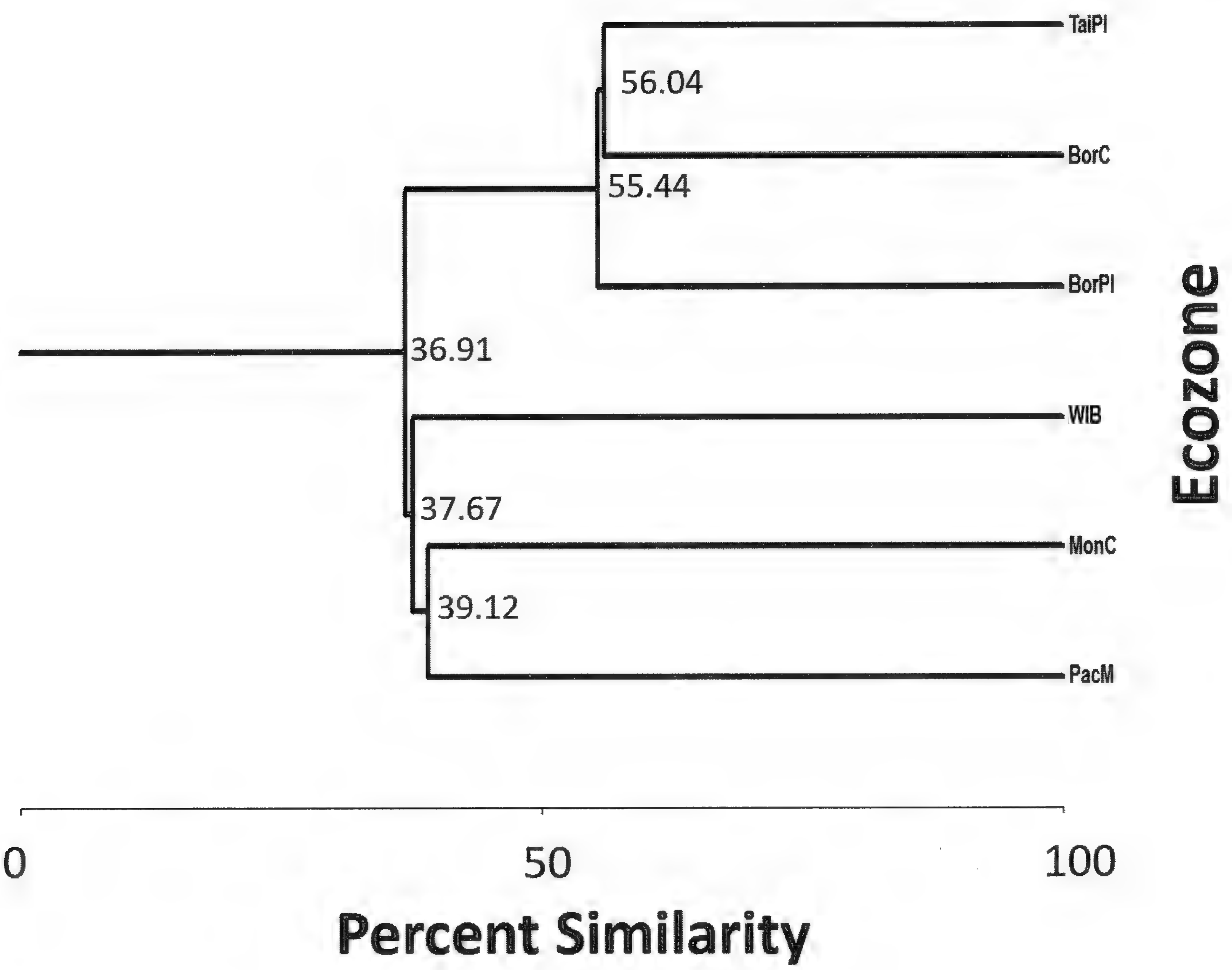

Figure 10. Incidence-based similarity (i.e., Jaccard's) of the bee fauna of each ecozone in British Columbia. The X-axis and numbers on the graph indicate percent similarity of each ecozone or group of ecozones, the right Y-axis indicates the ecozone: PacM=Pacific Maritime; WIB=Western Interior Basin; MonC=Montane Cordillera; BorPl=Boreal Plains; BorC=Boreal Cordillera; TaiPl=Taiga Plains.

ANNOTATED CHECKLIST OF THE BEES OF BRITISH COLUMBIA

	PacM	WIB	MonC	BorPl	BorC	TaiPl
FAMILY MELITTIDAE						
Subfamily Melittinae						
Genus *Macropis* Panzer, 1809						
Subgenus *Macropis* Panzer, 1809						
Macropis nuda (Provancher, 1882)	PacM	–	MonC	–	–	–

Species notes: Although Kline (2017) reported the family Melittidae (the genus *Macropis*) from British Columbia, presumably for the first time, specimens of *M. nuda* from Agassiz in the Canadian National Collection (Ottawa) were collected in 1914 (see Sheffield and Heron 2018). *Macropis nuda*, the likely species photographed by L.R. Best (see Kline 2017) based on the shiny terga (see Michez and Patiny 2005), is considered transcontinental (Snelling and Stage 1995) and is known to occur across most of southern Canada (Michez and Patiny 2005) and into Montana (Michener 1938a) in the United States. Michener (1938a) was the first to record the genus in western North America (presumably he did not examine material in the Canadian National Collection) – *M. nuda* (as *M. morsei* Robertson) from Colorado, and *M. steironematis* Robertson from Washington (Morgan's Ferry), Yakima River is the type locality for *Macropis steironematis*

opaca Michener, although Michener's subspecies is considered rare (Snelling and Stage 1995). It is possible that *M. steironematis* is also in the province.

FAMILY ANDRENIDAE
Subfamily Andreninae
***Andrena* Fabricius, 1775**
Subgenus *Andrena* Fabricius, 1775

Andrena aculeata LaBerge, 1980	–	WIB	MonC	–	–	–
Andrena buckelli Viereck, 1924	PacM	WIB	MonC	–	–	–
Andrena ceanothifloris cretata LaBerge, 1980	–	–	MonC	–	–	–
Andrena clarkella (Kirby, 1802)	–	–	MonC	BorPl	BorC	TaiPl
Andrena edwardsi Viereck, 1916	–	WIB	–	–	–	–
Andrena frigida Smith, 1853	PacM	WIB	MonC	BorPl	BorC	TaiPl
Andrena hemileuca Viereck, 1904	PacM	WIB	–	–	–	–
Andrena laminibucca Viereck & Cockerell, 1914	–	WIB	MonC	–	–	–
Andrena macoupinensis Robertson, 1900	–	WIB	–	–	–	–
Andrena milwaukeensis Graenicher, 1903	PacM	WIB	–	–	BorC	–
Andrena perarmata Cockerell, 1898	PacM	WIB	–	–	BorC	–
Andrena rufosignata Cockerell, 1902	PacM	WIB	MonC	BorPl	BorC	TaiPl
Andrena saccata Viereck, 1904	PacM	–	–	–	–	–
Andrena schuhi LaBerge, 1980	PacM	WIB	MonC	–	BorC	–
Andrena thaspii Graenicher, 1903	PacM	WIB	MonC	BorPl	BorC	TaiPl
Andrena topazana Cockerell, 1906	–	WIB	MonC	–	BorC	–
Andrena vicinoides Viereck, 1904	PacM	WIB	MonC	–	BorC	–
Andrena washingtoni Cockerell, 1901	PacM	WIB	MonC	–	BorC	–

Subgenus *Cnemidandrena* Hedicke, 1933

Andrena colletina Cockerell, 1906	–	WIB	–	–	–	–
Andrena columbiana Viereck, 1917	PacM	WIB	MonC	BorPl	BorC	TaiPl
**Andrena costillensis* Viereck & Cockerell, 1914	–	WIB	–	–	–	–
Andrena nubecula Smith, 1853	–	WIB	MonC	–	–	–
Andrena runcinatae Cockerell, 1906	–	–	MonC	–	–	–
Andrena scutellinitens Viereck, 1917	–	WIB	MonC	–	–	–
Andrena surda Cockerell, 1910	–	WIB	MonC	–	–	–

Species notes: Although Buckell (1949) reported *A. colletina* Cockerell from Chilcotin, Donovan (1977) indicated that the collection date (16 April 1921) was too early for this species; members of the subgenus *Cnemidandrena* are typically summer-flying species. However, Criddle (1924) examined specimens collected in September from Penticton and Cranbrook, so we include it in the list only from the WIB.

Subgenus *Dactylandrena* Viereck, 1924

Andrena berberidis Cockerell, 1905	–	WIB	–	–	–	–
Andrena porterae Cockerell, 1900	–	WIB	–	–	–	–

Subgenus *Dasyandrena* LaBerge, 1977

Andrena cristata Viereck, 1917	–	WIB	–	–	–	–

Subgenus *Diandrena* Cockerell, 1903

Andrena cuneilabris Viereck, 1926	–	WIB	–	–	–	–
Andrena evoluta Linsley & MacSwain, 1961	–	WIB	–	–	–	–
Andrena nothocalaidis Cockerell, 1905	–	WIB	–	–	–	–

Subgenus *Euandrena* Hedicke, 1933						
Andrena astragali Viereck & Cockerell, 1914	–	WIB	–	–	–	–
Andrena auricoma Smith, 1879	PacM	–	–	–	–	–
Andrena caerulea Smith, 1879	PacM	–	–	–	–	–
Andrena chlorura Cockerell, 1916	PacM	–	–	–	–	–
**Andrena geranii* Robertson, 1891	–	WIB	–	–	–	–
Andrena lawrencei Viereck & Cockerell, 1914	–	WIB	–	–	–	–
†*Andrena misella* Timberlake, 1951	–	WIB	–	–	–	–
Andrena nigrihirta (Ashmead, 1890)	PacM	WIB	MonC	BorPl	BorC	TaiPl
Andrena nigrocaerulea Cockerell, 1897	PacM	WIB	–	–	–	–
Andrena segregans Cockerell, 1900	–	–	MonC	–	–	–

Species notes: Although Linsley (1951b) reported *A. chlorura* Cockerell from the province, no specific details were provided. Ecozone information is provided from confirmed material at the Spencer Entomology Museum, University of British Columbia

Subgenus *Geissandrena* LaBerge & Ribble, 1972						
Andrena trevoris Cockerell, 1897	PacM	WIB	MonC	–	–	–
Subgenus *Holandrena* Pérez, 1890						
Andrena cressonii infasciata Lanham, 1949	PacM	WIB	–	–	–	–
Subgenus *Larandrena* LaBerge, 1964						
Andrena miserabilis Cresson, 1872	PacM	WIB	–	–	–	–
Subgenus *Leucandrena* Hedicke, 1933						
Andrena barbilabris (Kirby, 1802)	PacM	WIB	MonC	BorPl	BorC	TaiPl
Subgenus *Melandrena* Pérez, 1890						
Andrena carlini Cockerell, 1901	–	WIB	–	–	–	–
Andrena cerasifolii Cockerell, 1896	–	–	MonC	–	–	–
Andrena commoda Smith, 1879	–	WIB	–	–	–	–
Andrena lupinorum Cockerell, 1906	–	WIB	–	–	–	–
Andrena nivalis Smith, 1853	PacM	WIB	MonC	BorPl	BorC	TaiPl
Andrena pertristis Cockerell, 1905	–	WIB	MonC	–	–	–
Andrena regularis Malloch, 1917	–	WIB	MonC	BorPl	–	–
Andrena sola Viereck, 1917	–	WIB	–	–	–	–
Andrena transnigra Viereck, 1904	PacM	WIB	MonC	–	BorC	–
Andrena vicina Smith, 1853	PacM	WIB	MonC	–	–	–
Subgenus *Micrandrena* Ashmead, 1899						
Andrena candidiformis Viereck & Cockerell, 1914	–	WIB	–	–	–	–
Andrena chlorogaster Viereck, 1904	–	WIB	–	–	–	–
Andrena illinoiensis Robertson, 1891	–	WIB	–	–	–	–
Andrena melanochroa Cockerell, 1898	PacM	WIB	MonC	–	–	–
Andrena microchlora Cockerell, 1922	–	WIB	–	–	–	–
Andrena piperi Viereck, 1904	–	WIB	–	–	–	–
Andrena salictaria Robertson, 1905	–	WIB	MonC	BorPl	–	–
Subgenus *Parandrena* Robertson, 1897						
Andrena andrenoides (Cresson, 1878)	–	WIB	–	–	–	–
Andrena concinnula Cockerell, 1898	–	WIB	–	–	–	–

Andrena gibberis Viereck, 1924	–	WIB	–	–	–	–
Andrena nevadensis Cresson, 1879	–	WIB	–	–	–	–

Subgenus *Plastandrena* Hedicke, 1933

Andrena crataegi Robertson, 1893	PacM	WIB	–	–	–	–
Andrena prunorum prunorum Cockerell, 1896	PacM	WIB	MonC	BorP	–	–

Subgenus *Scaphandrena* Lanham, 1949

Andrena chapmanae Viereck, 1904	–	WIB	–	–	–	–
Andrena merriami Cockerell, 1901	–	WIB	–	–	–	–
Andrena montrosensis Viereck & Cockerell, 1914	–	WIB	MonC	–	–	–
Andrena scurra Viereck, 1904	PacM	WIB	MonC	–	–	–
Andrena sladeni Viereck, 1924	–	WIB	MonC	–	–	–
Andrena walleyi Cockerell, 1932	–	WIB	–	–	–	–

Species notes: Ribble (1974) considered *A. montrosensis* (recorded in the province by Buckell 1949) synonymous with a hybrid of *A. scurra* x *arabis* x *capricornis*, although Lanham (1984, 1987, 1993) later considered it a valid species, which is followed here.

Subgenus *Simandrena* Pérez, 1890

Andrena angustitarsata Viereck, 1904	PacM	WIB	MonC	–	–	–
Andrena pallidifovea Viereck, 1904	–	WIB	–	–	–	–
Andrena subtrita Cockerell, 1910	–	WIB	–	–	–	–
Andrena wheeleri Graenicher, 1904	–	–	MonC	BorPl	–	–

Subgenus *Thysandrena* Lanham, 1949

Andrena candida Smith, 1879	PacM	WIB	MonC	–	–	–
Andrena knuthiana Cockerell, 1901	–	WIB	MonC	–	–	–
Andrena medionitens Cockerell, 1902	PacM	WIB	MonC	–	–	–
Andrena trizonata (Ashmead, 1890)	–	WIB	–	–	–	–
Andrena vierecki Cockerell, 1904	PacM	–	–	–	–	–
Andrena w-scripta Viereck, 1904	PacM	WIB	MonC	BorPl	BorC	TaiPl

Subgenus *Trachandrena* Robertson, 1902

Andrena amphibola (Viereck, 1904)	PacM	WIB	–	–	–	–
Andrena cleodora (Viereck, 1904)	PacM	WIB	MonC	–	BorC	–
Andrena cupreotincta Cockerell, 1901	PacM	WIB	MonC	BorPl	–	–
Andrena cyanophila Cockerell, 1906	PacM	WIB	MonC	–	–	–
Andrena forbesii Robertson, 1891	PacM	WIB	MonC	BorPl	–	–
Andrena fuscicauda (Viereck, 1904)	–	WIB	–	–	–	–
Andrena hippotes Robertson, 1895	PacM	WIB	–	BorPl	–	TaiPl
Andrena mariae Robertson, 1891	PacM	WIB	MonC	BorPl	–	TaiPl
Andrena miranda Smith, 1879	PacM	WIB	MonC	BorPl	BorC	TaiPl
Andrena quintiliformis Viereck, 1917	–	WIB	–	–	–	–
Andrena salicifloris Cockerell, 1897	PacM	WIB	MonC	BorPl	–	–
Andrena sigmundi Cockerell, 1902	PacM	WIB	MonC	BorPl	–	TaiPl
Andrena striatifrons Cockerell, 1897	PacM	WIB	MonC	–	–	–

Subgenus *Tylandrena* LaBerge, 1964

Andrena erythrogaster (Ashmead, 1890)	PacM	–	–	–	–	–
Andrena perplexa Smith, 1853	PacM	–	MonC	–	–	–
Andrena subaustralis Cockerell, 1898	PacM	WIB	–	–	–	–
Andrena subtilis Smith, 1879	PacM	WIB	–	–	–	–

Unplaced Species

Andrena angustifovea Viereck, 1904	–	–	–	–	–	–

Andrena excellens Viereck, 1924	PacM	–	–	–	–	–
Andrena fulvicrista Viereck, 1924	PacM	WIB	–	–	–	–
Andrena lillooetensis Viereck, 1924	PacM	–	–	–	–	–
Andrena revelstokensis Viereck, 1924	–	–	MonC	–	–	–
Andrena singularis Viereck, 1924	PacM	–	MonC	–	–	–

Species notes: Though Viereck *et al.* (1904c) included *A. augustifovea* in their key to male *Andrena* in a treatment of bees of the Pacific North West, no specific information was provided in that work on the type material(s), including the number of specimens in the type series or the type locality. Cresson (1928) reviewed non-Cresson type material at the ANSP, including a specimen of *A. augustifovea* [ANSP no. 10,286] from Moscow, Idaho. Linsley (1951) and subsequent catalogues (i.e., Hurd 1979) have subsequently included British Columbia in the range of this species, suggesting other type material exists, even though we can find no further mention of this species in the literature. Although Linsley (1951) and Hurd (1979) did not assign this species to a subgenus, Ascher and Pickering (2018) place it within subgenus *Simandrena* Pérez and indicate three specimens (from Oregon, Idaho, and British Columbia); however, LaBerge (1989) did not include *A. angustifovea* as a valid species or as a synonymy in his revision of the subgenus. As such, we place it here until further work is done.

Subgenera not confirmed in British Columbia: Two male specimens identified by W. LaBerge as *Andrena* (*Anchandrena*) *angustella* Cockerell are in the Spencer Entomology Collection at the University of British Columbia – one from Vaseux Lake; the other from the north end of Galiano Island. Although LaBerge (1986) proposed and revised this subgenus, with this as the type species, both of the British Columbia specimens have an entirely black clypeus. A yellow clypeus (or yellow in part) is diagnostic for the subgenus (LaBerge 1986). As such, we consider these specimens misidentified. No specimens of *Anchandrena* have yet been reported from Canada (LaBerge 1986).
Criddle *et al.* (1924) reported *Andrena* (*Taeniandrena*) *wilkella* (Kirby) from British Columbia (Saanich), but this is well outside the known range of this introduced species establishment in North America. However, this could also represent another introduction event for this species in another major port area.

Subfamily Panurginae
Tribe Protandrini
Genus *Pseudopanurgus* Cockerell, 1897

Pseudopanurgus didirupa (Cockerell, 1908)	–	WIB	–	–	–	–

Tribe Panurgini
Genus *Panurginus* Nylander, 1848

**Panurginus atriceps* (Cresson, 1878)	–	WIB	MonC	–	BorC	–
†*Panurginus cressoniellus* Cockerell, 1898	–	WIB	–	–	–	–
**Panurginus ineptus* Cockerell, 1922	PacM	WIB	–	–	BorC	–

Tribe Perditini
Genus *Perdita* Smith, 1853
Subgenus *Perdita* Smith, 1853

Perdita fallax Cockerell, 1896	–	WIB	–	–	–	–

Subgenus *Pygoperdita* Timberlake, 1956

Perdita nevadensis Cockerell, 1896	PacM	WIB	–	–	–	–

Tribe Calliopsini
Genus *Calliopsis* Smith, 1853
Subgenus *Nomadopsis* Ashmead, 1898

Calliopsis scitula Cresson, 1878	–	WIB	–	–	–	–

FAMILY HALICTIDAE

Subfamily Rophitinae
Genus *Dufourea* Lepeletier, 1841

**Dufourea dilatipes* Bohart, 1948	–	WIB	MonC	–	–	–
Dufourea holocyanea (Cockerell, 1925)	–	WIB	MonC	–	–	–
Dufourea marginata (Cresson, 1878)	–	WIB	–	–	–	–
Dufourea maura (Cresson, 1878)	–	WIB	MonC	–	–	–
Dufourea trochantera Bohart, 1948	–	WIB	–	–	–	–

Other records: *Dufourea oryx* (Viereck) was recorded in British Columbia (Salmon Arm, Naramata) by Criddle *et al.* (1924), but it is likely that these are misidentified specimens of *D. holocyana.*

Subfamily Nomiinae
Genus *Nomia* Latreille, 1804
Subgenus *Acunomia* Cockerell, 1930

Nomia melanderi Cockerell, 1906	–	WIB	–	–	–	–

Species notes: This species was introduced to British Columbia (Ashcroft, Kamloops) from the western United States for alfalfa pollination (Bohart 1970; Hurd 1979), but there is no evidence that it became established in these areas. However, Stephen (1959) suggests that this species likely occurs naturally in southern parts of the interior valleys of the province.

Subfamily Halictinae
Tribe Halictini
Genus *Agapostemon* Guérin-Méneville, 1844
Subgenus *Agapostemon* Guérin-Méneville, 1844

Agapostemon femoratus Crawford, 1901	–	WIB	–	–	–	–
Agapostemon obliquus (Provancher, 1888)	PacM	–	–	–	–	–
Agapostemon texanus Cresson, 1872	PacM	WIB	MonC	–	–	–
Agapostemon virescens (Fabricius, 1775)	–	WIB	–	–	–	–

Genus *Halictus* Latreille, 1804
Subgenus *Nealictus* Pesenko, 1984

Halictus farinosus Smith, 1853	–	WIB	–	–	–	–

Subgenus *Odontalictus* Robertson, 1918

Halictus ligatus Say, 1837	–	WIB	–	–	–	–

Subgenus *Protohalictus* Pesenko, 1984

Halictus rubicundus (Christ, 1791)	PacM	WIB	–	–	–	TaiPl

Subgenus *Seladonia* Robertson, 1918

Halictus confusus arapahonum Cockerell, 1906	–	WIB	–	–	–	–
Halictus confusus confusus Smith, 1853	PacM	WIB	MonC	BorPl	–	–
Halictus tripartitus Cockerell, 1895	–	WIB	–	–	–	–
Halictus virgatellus Cockerell, 1901	–	WIB	–	–	–	TaiPl

Genus *Lasioglossum* Curtis, 1833
Subgenus *Dialictus* Robertson, 1902

Lasioglossum abundipunctum Gibbs, 2010	–	WIB	–	–	–	–
Lasioglossum albipenne (Robertson, 1890)	PacM	WIB	MonC	–	–	–

Lasioglossum albohirtum (Crawford, 1907)	PacM	WIB	MonC	–	–	–
Lasioglossum brunneiventre (Crawford, 1907)	PacM	WIB	–	–	–	–
Lasioglossum cressonii (Robertson, 1890)	PacM	WIB	MonC	BorPl	–	–
Lasioglossum dashwoodi Gibbs, 2010	–	WIB	–	–	–	–
Lasioglossum hyalinum (Crawford, 1907)	PacM	WIB	–	–	–	–
Lasioglossum imbrex Gibbs, 2010	–	WIB	–	–	–	–
Lasioglossum incompletum (Crawford, 1907)	PacM	WIB	MonC	–	–	–
Lasioglossum knereri Gibbs, 2010	PacM	WIB	MonC	–	–	–
Lasioglossum laevissimum (Smith, 1853)	PacM	WIB	MonC	BorPl	–	TaiPl
Lasioglossum lilliputense Gibbs, 2010	–	–	MonC	–	–	–
Lasioglossum macroprosopum Gibbs, 2010	–	WIB	–	–	–	–
Lasioglossum marinense (Michener, 1936)	PacM	WIB	–	–	–	–
Lasioglossum nevadense (Crawford, 1907)	PacM	WIB	–	–	–	–
Lasioglossum nigroviride (Graenicher, 1910)	–	WIB	MonC	BorPl	–	–
Lasioglossum novascotiae (Mitchell, 1960)	PacM	WIB	MonC	BorPl	BorC	TaiPl
†*Lasioglossum obnubilum* (Sandhouse, 1924)	–	–	MonC	–	–	–
Lasioglossum pacatum (Sandhouse, 1924)	PacM	WIB	–	–	–	–
Lasioglossum planatum (Lovell, 1905)	PacM	WIB	MonC	BorPl	–	TaiPl
Lasioglossum prasinogaster Gibbs, 2010	–	WIB	MonC	–	–	–
Lasioglossum pruinosum (Robertson, 1892)	PacM	WIB	MonC	–	–	–
Lasioglossum punctatoventre (Crawford, 1907)	–	WIB	–	–	–	–
Lasioglossum reasbeckae Gibbs, 2010	PacM	WIB	–	–	–	–
Lasioglossum ruidosense (Cockerell, 1897)	PacM	WIB	MonC	BorPl	BorC	TaiPl
Lasioglossum sagax (Sandhouse, 1924)	–	WIB	MonC	BorPl	–	–
Lasioglossum sandhousiellum Gibbs, 2010	PacM	WIB	–	–	–	–
Lasioglossum sedi (Sandhouse, 1924)	–	WIB	–	–	–	–
Lasioglossum subversans (Mitchell, 1960)	PacM	WIB	MonC	BorPl	–	–
Lasioglossum tenax (Sandhouse, 1924)	–	WIB	MonC	BorPl	BorC	TaiPl
Lasioglossum testaceum (Robertson, 1897)	–	WIB	–	–	–	–
Lasioglossum tuolumnense Gibbs, 2009	–	WIB	–	–	–	–
Lasioglossum yukonae Gibbs, 2010	PacM	–	–	–	BorC	–

Other records: *Lasioglossum atriventre* (Crawford) was declared *nomen dubium* by Gibbs (2010); the type locality is within British Columbia (Goldstream).

Subgenus *Evylaeus* Robertson, 1902						
†*Lasioglossum argemonis* (Cockerell, 1897)	PacM	WIB	–	–	–	–
Subgenus *Hemihalictus* Cockerell, 1897						
Lasioglossum diatretum (Vachal, 1904)	PacM	WIB	–	–	–	–
†*Lasioglossum glabriventre* (Crawford, 1907)	–	WIB	–	–	–	–
Lasioglossum inconditum (Cockerell, 1916)	PacM	WIB	MonC	BorPl	BorC	TaiPl
†*Lasioglossum kincaidii* (Cockerell, 1898)	–	WIB	–	–	–	–
Lasioglossum macoupinense (Robertson, 1895)	PacM	WIB	–	–	–	–
Lasioglossum ovaliceps (Cockerell, 1898)	PacM	WIB	–	–	–	–
Lasioglossum pectoraloides (Cockerell, 1895)	–	WIB	–	–	–	–
Subgenus *Lasioglossum* Curtis, 1833						
Lasioglossum anhypops McGinley, 1986	PacM	WIB	MonC	–	–	–
Lasioglossum athabascense (Sandhouse, 1933)	–	WIB	MonC	BorPl	–	TaiPl
Lasioglossum colatum (Vachal, 1904)	–	WIB	MonC	BorPl	–	–
Lasioglossum egregium (Vachal, 1904)	PacM	WIB	MonC	–	–	–
Lasioglossum mellipes (Crawford, 1907)	PacM	WIB	–	–	–	–
Lasioglossum olympiae (Cockerell, 1898)	PacM	–	–	–	–	–
Lasioglossum pacificum (Cockerell, 1898)	PacM	–	–	–	–	–
Lasioglossum sisymbrii (Cockerell, 1895)	PacM	WIB	MonC	–	–	–
Lasioglossum titusi (Crawford, 1902)	–	WIB	–	–	–	–
Lasioglossum trizonatum (Cresson, 1874)	–	WIB	MonC	–	–	–
Subgenus *Leuchalictus* Warncke, 1975						
Lasioglossum zonulum (Smith, 1848)	PacM	–	–	BorPl	–	–
Subgenus *Sphecodogastra* Ashmead, 1899						
Lasioglossum arctoum (Vachal, 1904)	–	–	MonC	–	–	–
Lasioglossum boreale Svensson, Ebmer & Sakagami, 1977	PacM	WIB	–	–	BorC	TaiPl
Lasioglossum comagenense (Knerer & Atwood, 1964)	–	–	MonC	–	BorC	–
Lasioglossum cooleyi (Crawford, 1906)	–	WIB	–	–	–	–
Lasioglossum cordleyi (Crawford, 1906)	PacM	–		–		–
Lasioglossum nigrum (Viereck, 1903)	PacM	–	–	–	–	–
Lasioglossum quebecense (Crawford, 1907)	PacM	–	MonC	–		–
Genus *Sphecodes* Latreille, 1804						

†*Sphecodes arvensiformis* Cockerell, 1904	PacM	WIB	–	–	–	–
**Sphecodes clematidis* Robertson, 1897	PacM	WIB	–	–	–	–
**Sphecodes pecosensis pecosensis* Cockerell, 1904	PacM	WIB	–	–	–	–
**Sphecodes prosphorus* Lovell & Cockerell, 1907	–	WIB	–	–	–	–
**Sphecodes solonis* Graenicher, 1911	–	WIB	–	–	–	TaiPl

FAMILY COLLETIDAE
Subfamily Colletinae
Tribe Colletini
***Colletes* Latreille, 1802**

Colletes compactus hesperius Swenk, 1906	–	WIB	–	–	–	–
Colletes consors pascoensis Cockerell, 1898	–	WIB	–	–	–	–
Colletes fulgidus fulgidus Swenk, 1904	PacM	WIB	MonC	–	–	–
Colletes gypsicolens Cockerell, 1897	–	WIB	–	–	–	–
Colletes hyalinus oregonensis Timberlake, 1951	–	WIB	–	–	–	–
Colletes impunctatus lacustris Swenk, 1906	–	WIB	–	BorPl	BorC	TaiPl
Colletes kincaidii Cockerell, 1898	PacM	WIB	MonC	–	–	–
Colletes mandibularis Smith, 1853	–	WIB	–	–	–	–
Colletes phaceliae Cockerell, 1906	–	WIB	–	–	BorC	–
Colletes simulans nevadensis Swenk, 1908	–	WIB	–	–	–	–
Colletes slevini Cockerell, 1925	–	WIB	–	–	–	–

Other records: As indicated by Stephen (1954), the record of *Colletes angelicus* Cockerell from British Columbia (Pentiction, Walhachin) by Criddle *et al.* (1924) is likely based on a misidentification, so is not included here.
The same is likely also true for *C. gilensis* Cockerell, recorded by Gibson (1914) (Similkameen, Okanagan), because the species distribution also seems restricted to the southern United States.

Subfamily Hylaeinae
***Hylaeus* Fabricius, 1793**
Subgenus *Cephalylaeus* Michener, 1942

Hylaeus basalis (Smith, 1853)	PacM	WIB	MonC	BorPl	–	TaiPl

Subgenus *Hylaeus* Fabricius, 1793

Hylaeus annulatus (Linnaeus, 1758)	PacM	WIB	MonC	BorPl	BorC	TaiPl
Hylaeus leptocephalus (Morawitz, 1871)	–	WIB	–	–	–	–
Hylaeus mesillae (Cockerell, 1896)	–	WIB	–	–	–	–
Hylaeus rudbeckiae (Cockerell & Casad, 1895)	–	WIB	–	–	–	–
Hylaeus verticalis (Cresson, 1869)	–	WIB	–	BorPl	–	–

Species notes: Elwell (2012) recorded *H. rudbeckia* from the Western Interior Basin, but this was not indicated in the follow-up publication (Elwell *et al.* 2016). This species was also recorded on Discover Life (Ascher and Pickering 2018) from material in the AMNH [Cache Creek].

Subgenus *Paraprosopis* Popov, 1939

Hylaeus coloradensis (Cockerell, 1896)	–	WIB	–	–	–	–

Hylaeus nevadensis (Cockerell, 1896)	–	WIB	–	–	–	–
Hylaeus wootoni (Cockerell, 1896)	–	WIB	–	–	–	–

Other records: Criddle *et al.* (1924) reported *H. cookii* (Metz) from British Columbia (Kaslo), but this was likely a misidentification; Snelling (1970) indicates that, until 1970, the species was known only from the type specimen (from New Mexico), and suggests that Metz's original description was not helpful for recognizing this species. Therefore, we do not include this species here.

Subgenus *Prosopis* Fabricius, 1804

Hylaeus affinis (Smith, 1853)	–	WIB	MonC	–	–	–
Hylaeus episcopalis (Cockerell, 1896)	–	WIB	–	–	–	–
Hylaeus modestus citrinifrons (Cockerell, 1896)	PacM	WIB	MonC	–	–	–

Species notes: Gibson and Criddle (1920) recorded *H. modestus* Say from British Columbia (Kaslo), but here we assume it was the subspecies *H. modestus citrinifrons*.

FAMILY MEGACHILIDAE
Subfamily Megachilinae
Tribe Osmiini
Genus *Ashmeadiella* Cockerell, 1897
Subgenus *Ashmeadiella* Cockerell, 1897

Ashmeadiella bucconis denticulata (Cresson, 1878)	–	WIB	–	–	–	–
Ashmeadiella cactorum cactorum (Cockerell, 1897)	–	WIB	–	–	–	–
Ashmeadiella californica californica (Ashmead, 1897)	–	WIB	–	–	–	–
Ashmeadiella cubiceps (Cresson, 1879)	–	WIB	–	–	–	–

Other records: Hurd and Michener (1955) showed a range map indicating that *Ashmeadiella* (*Argochila*) *foxiella* Michener was likely in British Columbia (Western Interior Basin), but no locality data were provided. Therefore, it is not included in the list above.

Genus *Atoposmia* Cockerell, 1935
Subgenus *Atoposmia* Cockerell, 1935

Atoposmia abjecta (Cresson, 1878)	–	–	MonC	–	–	–

Species notes: Hurd and Michener (1955) showed a range map indicating that *Atoposmia oregona* (Michener) was likely in southern British Columbia, but no locality data were provided. Therefore, it is not included in the list above.

Subgenus *Hexosmia* Michener, 1943

Atoposmia copelandica copelandica (Cockerell, 1908)	–	WIB	–	–	–	–

Genus *Chelostoma* Latreille, 1809
Subgenus *Foveosmia* Warncke, 1991

Chelostoma minutum Crawford, 1916	–	WIB	–	–	–	–
Chelostoma phaceliae Michener, 1938	–	WIB	–	–	–	–

Genus *Heriades* Spinola, 1808
Subgenus *Neotrypetes* Robertson, 1918

Heriades carinata Cresson, 1864	–	WIB	–	–	–	–
Heriades cressoni Michener, 1938	–	WIB	–	–	–	–
Heriades variolosa variolosa (Cresson, 1872)	–	WIB	–	–	–	–

Genus *Hoplitis* Klug, 1807
Subgenus *Alcidamea* Cresson, 1864

Hoplitis albifrons albifrons (Kirby, 1837)	–	–	MonC	BorPl	BorC	TaiPl
Hoplitis albifrons argentifrons (Cresson, 1864)	PacM	WIB	MonC	–	–	–
Hoplitis fulgida fulgida (Cresson, 1864)	PacM	WIB	MonC	BorPl	–	–
Hoplitis grinnelli septentrionalis Michener, 1947	–	WIB	MonC	–	–	–
Hoplitis hypocrita (Cockerell, 1906)	PacM	WIB	MonC	–	–	–
Hoplitis louisae (Cockerell, 1934)	PacM	WIB	MonC	–	–	–
Hoplitis producta subgracilis Michener, 1947	PacM	WIB	MonC	–	–	–
Hoplitis sambuci Titus, 1904	PacM	WIB	MonC	–	–	–
Hoplitis spoliata (Provancher, 1888)	–	WIB	MonC	BorPl	–	TaiPl

Species notes: Michener (1947b) and Hurd and Michener (1955) indicate that *H. albifrons albifrons* occurrs across Canada, including in northern British Columbia, being replaced by *H. albifrons argentifrons* in the southern part of the province. Michener (1947a) indicates that separation of the subspecies (based on hair colour) in some areas would likely be difficult, but DNA barcoding suggests there is much variation within this species in the province (i.e., three clusters with no apparent geographic pattern) all sharing a single Barcode Index Number. Incidentally, there are three subspecies in North America (Michener 1947a, b; Hurd and Michener 1955; Rowe 2017).

Subgenus *Formicapis* Sladen, 1916

Hoplitis robusta robusta (Nylander, 1848)	PacM	WIB	MonC	BorPl	BorC	TaiPl

Species notes: Michener (1938c) recorded this species from MonC (Field); Hurd (1979) recorded this Holarctic species from British Columbia, but no specific localities were provided. However, it is likely found in all ecozones in the province.

Genus *Osmia* Panzer, 1806
Subgenus *Cephalosmia* Sladen, 1916

Osmia californica Cresson, 1864	–	WIB	–	–	–	–
Osmia marginipennis Cresson, 1878	–	WIB	–	–	–	–
Osmia montana montana Cresson, 1864	–	WIB	MonC	–	–	–
Osmia subaustralis Cockerell, 1900	–	–	MonC	BorPl	–	–

Subgenus *Helicosmia* Thomson, 1872

Osmia caerulescens caerulescens (Linnaeus, 1758)	PacM	WIB	MonC	–	–	–
Osmia coloradensis Cresson, 1878	–	WIB	MonC	–	–	–
Osmia texana Cresson, 1872	PacM	WIB	–	–	–	–

Subgenus *Melanosmia* Schmiedeknecht, 1885

Osmia albolateralis Cockerell, 1906	PacM	WIB	MonC	–	–	–
**Osmia aquilonaria* Rightmyer, Griswold & Arduser, 2010	–	–	MonC	–	–	–
**Osmia atriventris* Cresson, 1864	–	–	MonC	–	–	–
Osmia atrocyanea Cockerell, 1897	–	WIB	–	–	–	–
Osmia austromaritima Michener, 1936	–	WIB	–	–	–	–

Osmia bella Cresson, 1878	PacM	WIB	MonC	–	–	–
Osmia brevis brevis Cresson, 1864	PacM	WIB	–	–	–	–
Osmia bruneri Cockerell, 1897	–	WIB	MonC	–	–	–
Osmia bucephala Cresson, 1864	PacM	WIB	MonC	BorPl	BorC	TaiPl
Osmia calla Cockerell, 1897	–	WIB	–	–	–	–
Osmia cobaltina Cresson, 1878	–	WIB	–	–	–	–
Osmia cyanella Cockerell, 1897	–	WIB	–	–	–	–
Osmia cyaneonitens Cockerell, 1906	–	WIB	–	–	–	–
Osmia densa densa Cresson, 1864	PacM	WIB	MonC	–	–	–
Osmia dolerosa Sandhouse, 1939	PacM	WIB	–	–	–	–
Osmia ednae Cockerell, 1907	–	WIB	–	–	–	–
Osmia enixa Sandhouse, 1924	–	WIB	–	–	–	–
Osmia exigua Cresson, 1878	–	WIB	–	–	–	–
Osmia giliarum Cockerell, 1906	–	WIB	MonC	–	–	–
Osmia inermis (Zetterstedt, 1838)	–	–	MonC	BorPl	BorC	TailPl
Osmia integra Cresson, 1878	–	WIB	MonC	–	–	TaiPl
Osmia inurbana Cresson, 1878	PacM	WIB	–	–	–	–
Osmia juxta juxta Cresson, 1864	–	WIB	MonC	–	–	–
Osmia juxta subpurpurea Cockerell, 1897	–	WIB	–	–	–	–
Osmia kincaidii Cockerell, 1897	PacM	WIB	MonC	–	–	–
†*Osmia laeta* Sandhouse, 1924	–	WIB	MonC	–	–	–
Osmia lignaria propinqua Cresson, 1864	PacM	WIB	MonC	–	–	–
Osmia longula Cresson, 1864	–	WIB	MonC	–	BorC	–
†*Osmia malina* Cockerell, 1909	–	WIB	–	–	–	–
Osmia melanopleura Cockerell, 1916	–	WIB	–	–	–	–
Osmia mertensiae Cockerell, 1907	PacM	WIB	–	–	–	–
Osmia nanula Cockerell, 1897	PacM	WIB	–	–	–	–
Osmia nemoris Sandhouse, 1924	–	WIB	–	–	–	–
Osmia nifoata Cockerell, 1909	–	WIB	–	–	–	–
Osmia nigrifrons Cresson, 1878	–	WIB	MonC	–	–	–
Osmia nigriventris (Zetterstedt, 1838)	PacM	WIB	MonC	BorPl	BorC	–
Osmia obliqua White, 1952	–	WIB	–	–	–	–
Osmia odontogaster Cockerell, 1897	–	WIB	MonC	–	–	–
**Osmia paradisica* Sandhouse, 1924	–	WIB	–	–	–	–
Osmia pentstemonis Cockerell, 1906	–	WIB	–	–	–	–
Osmia pikei Cockerell, 1907	–	WIB	–	–	–	–
Osmia proxima Cresson, 1864	PacM	WIB	MonC	BorPl	BorC	TaiPl
†*Osmia pulsatillae* Cockerell, 1907	–	WIB	MonC	–	–	–
Osmia pusilla Cresson, 1864 PacM	–	WIB	–	–	–	–
†*Osmia raritatis* Michener, 1957	–	WIB	–	–	–	–
Osmia regulina Cockerell, 1911	–	WIB	–	–	–	–
Osmia sedula Sandhouse, 1924	–	WIB	–	–	–	–
Osmia simillima Smith, 1853	–	WIB	MonC	BorPl	–	–
Osmia tersula Cockerell, 1912	–	–	MonC	BorPl	–	–
Osmia trevoris Cockerell, 1897	–	WIB	–	–	–	–
Osmia tristella tristella Cockerell, 1897	PacM	WIB	–	BorPl	–	–
Osmia unca Michener, 1937	–	WIB	–	–	–	–

Species notes: *Osmia mertensiae* Cockerell and *Osmia inurbana* Cresson (as *O. eutrichosa* Cockerell) were recorded from British Columbia by Sandhouse (1925b) so are listed here, but Hurd (1979) considers the species questionable from British Columbia.

Tribe Anthidiini
Genus *Anthidiellum* Cockerell, 1904
Subgenus *Loyolanthidium* Urban, 2001

Anthidiellum robertsoni	–	WIB	–	–	–	–

(Cockerell, 1904)

Species notes: Based on distinct differences in the cytochrome c oxidase I (COI) gene that resulted in two distinct Barcode Index Numbers (BINs) (see Sheffield *et al.* 2017), we agree with Urban (2001) and consider this a separate species from the eastern *A. notatum* (Latreille).

Genus *Anthidium* Fabricius, 1804
Subgenus *Anthidium* Fabricius, 1804

Anthidium atrifrons Cresson, 1868	–	WIB	–	–	–	–
Anthidium clypeodentatum Swenk, 1914	–	WIB	MonC	–	–	TaiPl
Anthidium emarginatum (Say, 1824)	–	WIB	MonC	–	–	–
†*Anthidium formosum* Cresson, 1878	–	WIB	–	–	–	–
Anthidium manicatum (Linnaeus, 1758)	PacM	–	MonC	–	–	–
Anthidium mormonum Cresson, 1878	–	WIB	MonC	–	–	–
Anthidium palliventre Cresson, 1878	–	–	MonC	–	–	–
Anthidium psoraleae Robertson, 1902	–	WIB	–	–	–	–
Anthidium tenuiflorae Cockerell, 1907	–	WIB	–	–	–	TaiPl
Anthidium utahense Swenk, 1914	–	WIB	MonC	–	–	–

Species notes: Although Michener (1951) and Hurd (1979) recorded *A. porterae* Cockerell from "BC" (no specific locality), we have found reference to this species in Canada only from Alberta (Calgary) by Cockerell (1912). Gonzalez and Griswold (2013) and Griswold *et al.* (2014) did not record this species from Canada in their revision and compilation of occurrence records for the genus in the Western Hemisphere, respectively.

Genus *Dianthidium* Cockerell, 1900
Subgenus *Dianthidium* Cockerell, 1900

Dianthidium curvatum sayi Cockerell, 1907	–	WIB	–	–	–	–
†*Dianthidium plenum plenum* Timberlake, 1943	–	WIB	–	–	–	–
Dianthidium pudicum pudicum (Cresson, 1879)	–	WIB	–	–	–	–
†*Dianthidium singulare* (Cresson, 1879)	–	WIB	–	–	–	–
Dianthidium subparvum Swenk, 1914	PacM	WIB	–	–	–	–
Dianthidium ulkei ulkei (Cresson, 1878)	–	WIB	–	–	–	–

Genus *Stelis* Panzer, 1806
Subgenus *Stelis* Panzer, 1806

†*Stelis ashmeadiellae* Timberlake, 1941	PacM	–	–	–	–	–
†*Stelis calliphorina* (Cockerell, 1911)	–	WIB	–	–	–	–
Stelis callura Cockerell, 1925	–	WIB	–	–	–	–
Stelis carnifex Cockerell, 1911	–	WIB	–	–	–	–
Stelis coarctatus Crawford, 1916	–	WIB	–	–	–	–
Stelis elegans Cresson, 1864	–	WIB	–	–	–	–
Stelis lateralis Cresson, 1864	PacM	–	–	–	–	–
Stelis maculata (Provancher, 1888)	PacM	–	–	–	–	–
Stelis montana Cresson, 1864	–	WIB	–	–	–	–
Stelis monticola Cresson, 1878	–	WIB	–	–	–	–
Stelis occidentalis Parker & Griswold, 2013	–	WIB	–	–	–	–
Stelis ricardonis (Cockerell, 1912)	PacM	WIB	–	–	–	–
Stelis rubi Cockerell, 1898	–	WIB	MonC	–	–	–

Tribe Dioxyini
Genus *Dioxys* Lepeletier & Serville, 1825

†*Dioxys pomonae pomonae* Cockerell, 1910	–	WIB	–	–	–	–

Species notes: This is the first record of this species from Canada; however, Sheffield *et al.* (2017) recorded this genus (this species, based on this single barcoded specimen) from British Columbia, Canada.

Tribe Megachilini
Genus *Coelioxys* Latreille, 1809
Subgenus *Boreocoelioxys* Mitchell, 1973

Coelioxys banksi Crawford, 1914	PacM	WIB	MonC	–	–	–
Coelioxys funeraria Smith, 1854	PacM	WIB	–	BorPl	BorC	TailPl
Coelioxys moesta Cresson, 1864	PacM	WIB	MonC	BorPl	BorC	TaiPl
Coelioxys novomexicana Cockerell, 1909	–	WIB	–	–	–	–
Coelioxys octodentata Say, 1824	–	WIB	MonC	–	–	–
Coelioxys porterae Cockerell, 1900	PacM	WIB	MonC	BorPl	–	–
Coelioxys rufitarsis Smith, 1854	PacM	WIB	MonC	–	–	TaiPl
Coelioxys sayi Robertson, 1897	–	WIB	–	–	–	–

Subgenus *Coelioxys* Latreille, 1809

Coelioxys hirsutissima Cockerell, 1912	–	WIB	–	–	–	–
Coelioxys sodalis Cresson, 1878	PacM	WIB	MonC	–	BorC	TaiPl

Subgenus *Cyrtocoelioxys* Mitchell, 1973

Coelioxys deani Cockerell, 1909	–	WIB	–	–	–	–

Subgenus *Synocoelioxys* Mitchell, 1973

Coelioxys alternata Say, 1837	PacM	WIB	MonC	–	–	–
Coelioxys apacheorum Cockerell, 1900	PacM	WIB	–	–	–	–
Coelioxys erysimi Cockerell, 1912	–	WIB	MonC	–	–	–

Subgenus *Xerocoelioxys* Mitchell, 1973

Coelioxys edita Cresson, 1872	–	WIB	–	–	–	–
Coelioxys grindeliae Cockerell, 1900	PacM	WIB	–	–	–	–

Genus *Megachile* Latreille, 1802
Subgenus *Argyropile* Mitchell, 1934

Megachile parallela Smith, 1853	PacM	WIB	MonC	–	–	–

Subgenus *Chelostomoides* Robertson, 1901

Megachile angelarum Cockerell, 1902	PacM	WIB	–	–	–	–

Species notes: Although *Megachile* (*Chelostomoides*) *subexilis* Cockerell was recorded from British Columbia (Kaslo, Penticton) by Gibson (1917), it is likely that this is based on misidentified specimens. Gibson (1917) reported this species in both Ontario and British Columbia, but he likely confused it with *M. campanulae* (Robertson) and *M. angelicus* found in each of those provinces, respectively (see Sheffield *et al.* 2011). Interestingly, Criddle *et al.* (1924) also record it from Alberta, Saskatchewan, Manitoba, and Fort Norman (Northwest Territories), supporting that these records were misidentified.

Subgenus *Eutricharaea* Thomson, 1872

Megachile apicalis Spinola, 1808	PacM	WIB	–	–	–	–

Megachile rotundata (Fabricius, 1793)	PacM	WIB	–	–	–	–
Subgenus *Litomegachile* Mitchell, 1934						
Megachile brevis Say, 1837	–	WIB	–	–	–	–
Megachile cleomis Cockerell, 1900	–	WIB	–	–	–	–
Megachile coquilletti Cockerell, 1915	–	WIB	–	–	–	–
Megachle gentilis Cresson, 1872	PacM	WIB	–	–	–	–
Megacile lippiae Cockerell, 1900	–	WIB	–	–	–	–
Megachle mendica Cresson, 1878	–	WIB	–	–	–	–
Megachile onobrychidis Cockerell, 1908	–	WIB	–	–	–	–
Megachile snowi Mitchell, 1927	–	WIB	–	–	–	–
Megachile texana Cresson, 1878	PacM	WIB	–	–	–	–
Subgenus *Megachile* Latreille, 1802						
Megachile centuncularis (Linnaeus, 1758)	PacM	WIB	MonC	–	–	–
Megachile inermis Provancher, 1888	–	WIB	MonC	BorPl	–	–
Megachile lapponica Thomson, 1872	PacM	–	MonC	BorPl	BorC	TaiPl
Megachile montivaga Cresson, 1878	PacM	WIB	MonC	–	–	–
Megachile relativa Cresson, 1878	PacM	WIB	MonC	BorPl	BorC	TaiPl
Subgenus *Megachiloides* Mitchell, 1924						
Megachile subnigra Cresson, 1879	–	WIB	–	–	–	–
Megachile umatillensis (Mitchell, 1927)	–	WIB	–	–	–	–
Megachine wheeleri Mitchell, 1927	–	WIB	–	–	–	–
Subgenus *Sayapis* Titus, 1906						
Megachile fidelis Cresson, 1878	PacM	WIB	–	–	–	–
Megachile mellitarsis Cresson, 1878	–	WIB	–	–	–	–
†*Megachile pugnata pomonae* Cockerell, 1916	–	WIB	–	–	–	–
Megachile pugnata pugnata Say, 1837	PacM	WIB	MonC	BorPl	–	TaiPl
Subgenus *Xanthosarus* Robertson, 1903						
Megachile circumcincta (Kirby, 1802)	–	WIB	MonC	BorPl	BorC	TaiPl
Megachile frigida Smith, 1853	PacM	WIB	MonC	BorPl	BorC	TaiPl
Megachile gemula Cresson, 1878	PacM	WIB	MonC	BorPl	–	TaiPl
Megachile melanophaea Smith, 1853	PacM	WIB	MonC	BorPl	BorC	TaiPl
Megachile perihirta Cockerell, 1898	PacM	WIB	MonC	BorPl	BorC	TaiPl

Subgenera not confirmed in British Columbia: Criddle *et al.* (1924) recorded *Megachile* (*Pseudocentron*) *pruina* Smith from western Canada (including Summerland, British Columbia), but this subgenus has not been recorded in Canada (Sheffield *et al.* 2011). It is suspected these records are misidentified specimens of *M. parallela* Smith.

FAMILY APIDAE
Subfamily Xylocopinae
Tribe Ceratinini
Genus *Ceratina* Latreille, 1802
Subgenus *Zadontomerus* Ashmead, 1899

Ceratina acantha Provancher, 1895	PacM	WIB	MonC	–	–	–
Ceratina nanula Cockerell, 1897	PacM	WIB	MonC	–	–	–
Ceratina pacifica Smith, 1907	PacM	WIB	–	–	–	–

Subfamily Nomadinae
Tribe Nomadini

Genus *Nomada* Scopoli, 1770

Nomada aldrichi Cockerell, 1910	–	WIB	–	–	–	–
Nomada articulata Smith, 1854	–	–	MonC	–	–	–
Nomada bella Cresson, 1863	PacM	WIB	MonC	–	–	–
Nomada citrina Cresson, 1878	PacM	–	–	–	–	–
Nomada civilis Cresson, 1878	–	WIB	–	–	–	–
Nomada corvallisensis Cockerell, 1903	–	WIB	–	–	–	–
†*Nomada crotchii* Cresson, 1878	PacM	WIB	–	–	–	–
Nomada edwardsii Cresson, 1878	PacM	WIB	–	–	–	–
Nomada grayi Cockerell, 1903	PacM	–	–	–	–	–
Nomada pascoensis Cockerell, 1903	–	WIB	–	–	–	–
Nomada perbella (Viereck, 1905)	PacM	–	–	–	–	–
Nomada rhodomelas Cockerell, 1903	PacM	–	–	–	–	–
Nomada sayi Robertson, 1893	–	WIB	–	–	–	–
Nomada scita Cresson, 1878	–	WIB	–	–	–	–
Nomada superba Cresson, 1863	PacM	–	MonC	–	–	–
**Nomada texana* Cresson, 1872	–	WIB	–	–	–	–
Nomada ultima Cockerell, 1903	PacM	–	–	–	–	–
Nomada valida Smith, 1854	–	WIB	–	–	–	–
Nomada vernonensis Cockerell, 1916	–	WIB	–	–	–	–

Species notes: *Nomada proxima* Cresson was recorded from British Columbia (Vernon) by Viereck (1926), but that species is known only from type material from Maine. We presume Viereck's record to be misidentified.
Mitchell (1962) and Hurd (1979) recorded *Nomada valida* Smith from British Columbia, but provided no specific localities. However, *N. nigrocincta* Smith, recorded from Penticton by Criddle *et al.* (1924), is considered an unpublished synonymy of *N. valida* (opinion of Snelling, as cited by Ascher and Pickering 2018).

Tribe Epeolini
Genus *Epeolus* Latreille, 1802

Epeolus americanus (Cresson, 1878)	–	WIB	MonC	–	–	–
Epeolus compactus Cresson, 1878	–	WIB	MonC	–	–	–
Epeolus minimus (Robertson, 1902)	PacM	WIB	MonC	–	BorC	–
Epeolus olympiellus Cockerell, 1904	PacM	WIB	–	–	–	–

Genus *Triepeolus* Robertson, 1901

Triepeolus occidentalis (Cresson, 1878)	–	WIB	–	–	–	–
Triepeolus paenepectoralis Viereck, 1905	PacM	–	–	–	–	–
Triepeolus subalpinus Cockerell, 1910	–	WIB	–	–	–	–
Triepeolus texanus (Cresson, 1878)	–	WIB	–	–	–	–

Tribe Biastini
Genus *Neopasites* Ashmead, 1898

Neopasites aff. *fulviventris* (Cresson, 1878)	–	WIB	–	–	–	–

Tribe Emphorini
Genus *Diadasia* Patton, 1879

**Diadasia australis* (Cresson, 1878)	–	WIB	MonC	–	–	–
Diadasia diminuta (Cresson, 1878)	–	WIB	MonC	–	–	–

Tribe Eucerini
Genus *Eucera* Scopoli, 1770
Subgenus *Synhalonia* Patton, 1879

Eucera acerba (Cresson, 1879)	–	WIB	–	–	–	–

Eucera actuosa (Cresson, 1878)	PacM	WIB	–	–	–	–
Eucera cordleyi (Viereck, 1905)	–	WIB	–	–	–	–
Eucera douglasiana (Cockerell, 1906)	–	WIB	–	–	–	–
Eucera edwardsii (Cresson, 1878)	–	WIB	–	–	–	–
Eucera frater lata (Provancher, 1888)	PacM	–	–	–	–	–
Eucera fulvitarsis fulvitarsis (Cresson, 1878)	–	WIB	–	–	–	–
Eucera hirsutissima (Cockerell, 1916)	PacM	–	–	–	–	–
Eucera hurdi (Timberlake, 1969)	–	–	–	–	–	–
Eucera virgata (Cockerell, 1905)	–	WIB	–	–	–	–

Species notes: *Eucera hirsutissima* (Cockerell) was recorded from "British Columbia" by Cockerell (1916b), Gibson (1918), and Hurd (1979) though no specific localities were provided. He (Cockerell 1916b) indicated that a second label, "Toba" was on the type specimen at the British Museum, suggesting Toba Inlet on Powell River. Thus, we include the PacM in the list above. Similarly, *E. hurdi* was recorded from the province by Hurd (1979), but no other literature records are known. Thus, we do not specify ecozone information for this species.

Genus *Melissodes* Latreille, 1825
Subgenus *Eumelissodes* LaBerge, 1956

Melissodes agilis Cresson, 1878	–	–	MonC	–	–	–
Melissodes bimatris LaBerge, 1961	–	WIB	–	–	–	–
Melissodes lutulentus LaBerge, 1961	–	WIB	–	–	–	–
Melissodes menuachus Cresson, 1868	–	WIB	–	–	–	–
Melissodes microstictus Cockerell, 1905	PacM	WIB	–	–	–	–
Melissodes pallidisignatus Cockerell, 1905	–	WIB	–	–	–	–
†*Melissodes saponellus* Cockerell, 1908	–	WIB	–	–	–	–
Melissodes semilupinus Cockerell, 1905	–	WIB	–	–	–	–

Species notes: Although Michener (1951e) recorded *Melissodes illatus* Lovell and Cockerell from the province, no additional information was provded. However, LaBerge (1961) did not record it from British Columbia in his revision, so it is not included here.

Subgenus *Heliomelissodes* LaBerge, 1956

Melissodes rivalis Cresson, 1872	–	WIB	–	–	–	–

Subgenus *Melissodes* Latreille, 1825

Melissodes communis alopex Cockerell, 1928	–	WIB	–	–	–	–

Tribe Anthophorini
Genus *Anthophora* Latreille, 1803
Subgenus *Clisodon* Patton, 1879

Anthophora terminalis Cresson, 1869	PacM	WIB	MonC	BorPl	BorC	TaiPl

Subgenus *Lophanthophora* Brooks, 1988

Anthophora pacifica Cresson, 1878	–	WIB	–	–	–	–
Anthophora porterae Cockerell, 1900	PacM	WIB	–	–	–	–
Anthophora ursina Cresson, 1869	–	WIB	MonC	–	–	–

Subgenus *Melea* Sandhouse, 1943

Anthophora bomboides Kirby, 1838	PacM	WIB	MonC	BorPl	BorC	TaiPl
Anthophora occidentalis Cresson, 1869	PacM	WIB	–	–	–	–

Subgenus *Micranthophora* Cockerell, 1906

Anthophora peritomae Cockerell, 1905	–	WIB	–	–	–	–

Subgenus *Mystacanthophora* Brooks, 1988

**Anthophora urbana* Cresson, 1878	PacM	WIB	–	–	–	–

Subgenus *Pyganthophora* Brooks, 1988

Anthophora crotchii Cresson, 1878	–	WIB	–	–	–	–
Anthophora edwardsii Cresson, 1878	–	WIB	MonC	–	–	–

Genus *Habropoda* Smith, 1854

Habropoda cineraria (Smith, 1879)	PacM	WIB	–	–	–	–
†*Habropoda miserabilis* (Cresson, 1878)	PacM	–	–	–	–	–
Habropoda murihirta (Cockerell, 1905)	–	WIB	–	–	–	–

Species notes: Stainer (1959) and Hurd (1979, likely based on Stainer's publication) recorded *Habropoda murihirta* (Cockerell) from British Columbia, likely Okanagan Mission. We have not been able to locate this material (17 specimens) in the CNC and, although we assume that these were likely specimens of *H. cineraria*, we leave it in the list.

Tribe Melectini
Genus *Melecta* Latreille, 1802
Subgenus *Melecta* Latreille, 1802

Melecta pacifica fulvida Cresson, 1878	PacM	WIB	MonC	–	–	–
Melecta pacifica pacifica Cresson, 1878	PacM	WIB	–	–	–	–
Melecta separata callura (Cockerell, 1926)	–	WIB	–	–	–	–
Melecta thoracica Cresson, 1875	–	WIB	MonC	–	–	–

Genus *Xeromelecta* Linsley, 1939
Subgenus *Melectomorpha* Linsley, 1939

Xeromelecta californica (Cresson, 1878)	–	WIB	–	–	–	–

Tribe Bombini
Genus *Bombus* Latreille, 1802
Subgenus *Alpinobombus* Skorikov, 1914

Bombus kirbiellus Curtis, 1835	–	–	MonC	BorPl	BorC	TaiPl
Bombus neoboreus Sladen, 1919	PacM	–	MonC	–	BorC	TaiPl
Bombus polaris Curtis, 1835	–	–	MonC	–	BorC	TalPl

Species notes: Although *B. natvigi* Richards (= North American *B. hyperboreus* Schönherr) was listed from "British Columbia" by Cannings (2011), no records were recorded by Williams *et al.* (2014; as *B. hyperboreus*). Although it is likely that this species does occur at high elevations and/or latitudes in the province, we have not yet found any records supporting this, so we do not include it here.

Subgenus *Bombias* Robertson, 1903

Bombus nevadensis Cresson, 1874	PacM	WIB	MonC	BorPl	BorC	–

Subgenus *Bombus* Latreille, 1802

Bombus cryptarum (Fabricius, 1775)	–	WIB	MonC	BorPl	BorC	TaiPl
Bombus occidentalis mckayi Ashmead, 1902	–	–	–	–	BorC	–

Bombus occidentalis occidentalis Greene, 1858	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus terricola Kirby, 1837	–	WIB	MonC	BorPl	BorC	TaiPl

Subgenus *Cullumanobombus* Vogt, 1911

Bombus griseocollis (DeGeer, 1773)	–	WIB	–	–	–	–
Bombus morrisoni Cresson, 1878	PacM	WIB	–	–	–	–
Bombus rufocinctus Cresson, 1863	PacM	WIB	MonC	BorPl	–	–

Subgenus *Psithyrus* Lepeletier, 1833

Bombus bohemicus (Seidl, 1837)	–	WIB	MonC	–	BorC	TaiPl
Bombus flavidus Eversmann, 1852	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus insularis (Smith, 1861)	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus suckleyi Greene, 1860	PacM	WIB	MonC	BorPl	BorC	TaiPl

Subgenus *Pyrobombus* Dalla Torre, 1880

Bombus bifarius Cresson, 1878	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus caliginosus (Frison, 1927)	PacM	–	–	–	–	–
Bombus centralis Cresson, 1864	PacM	WIB	MonC	BorPl	BorC	–
Bombus flavifrons Cresson, 1863	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus frigidus Smith, 1854	–	–	MonC	BorPl	BorC	TaiPl
Bombus huntii Greene, 1860	–	WIB	MonC	BorPl	–	–
Bombus impatiens Cresson, 1863	PacM	–	–	–	–	–
Bombus jonellus (Kirby, 1802)	–	–	MonC	–	BorC	TaiPl
Bombus melanopygus Nylander, 1848	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus mixtus Cresson, 1878	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus perplexus Cresson, 1863	PacM	WIB	–	BorPl	BorC	–
Bombus sitkensis Nylander, 1848	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus sylvicola Kirby, 1837	PacM	WIB	MonC	BorPl	BorC	TaiPl
Bombus ternarius Say, 1837	–	–	MonC	BorPl	–	–
Bombus vagans vagans Smith, 1854	–	WIB	–	–	–	–
Bombus vandykei (Frison, 1927)	–	WIB	MonC	–	–	–
Bombus vosnesenskii Radoszkowski, 1862	PacM	WIB	MonC	–	–	–

Species notes: *Bombus impatiens* was first recorded as an established species in British Columbia by Ratti and Colla (2010; but see Ratti 2006), but it has been used as a commercial pollinator in the province for much longer (see Van Westendorp and McCutcheon 2001).

Although *B. sandersoni* Franklin was recorded from British Columbia by Williams *et al.* (2014), it is likely that this specimen is misidentified, and is thus removed from the provincial list until it can be confirmed.

Subgenus *Subterraneobombus* Vogt, 1911

Bombus appositus Cresson, 1878	PacM	WIB	MonC	–	–	–
Bombus borealis Kirby, 1837	–	–	–	BorPl	–	TaiPl

Subgenus *Thoracobombus* Dalla Torre, 1880

Bombus fervidus (Fabricius, 1798)	PacM	WIB	MonC	BorPl	BorC	–

Other records: Venables (1914) recorded *Bombus pensylvanicus* (De Geer) from British Columbia, but it is likely that these specimen(s) were of the dark form of *B. nevadensis* or possibly *B. terricola* (see Williams *et al.* 2014). Earlier authors (e.g., Viereck *et al.* 1904a) considered this name synonymous with *B. fervidus*. During research for a recent Committee on the Status of Endangered Wildlife in Canada (COSEWIC) assessment of *B. pensylvanicus* in Canada, all records from west of southern Ontario in Canada were found to be misidentified (C.S.S., unpublished). Stephen (1957) did not record *B. pensylvanicus* (as *B. sonorus* Say) from British Columbia. Similarly, Buckell [1951; and later Hurd (1979) and Cannings (2011)] recorded *Bombus auricomus* Robertson from British Columbia (Centurian, and Departure Bay [Vancouver Island]), but it is

likely that these specimens and possibly other specimens of this species recorded from western Canada are the dark form of *B. nevadensis*.

Tribe Apini
Genus *Apis* Linnaeus, 1758
Subgenus *Apis* Linnaeus, 1758

Apis mellifera Linnaeus, 1758	PacM	WIB	MonC	BorPl	BorC	–

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Efficacy of diamide, neonicotinoid, pyrethroid, and phenyl pyrazole insecticide seed treatments for controlling the sugar beet wireworm, *Limonius californicus* (Coleoptera: Elateridae), in spring wheat

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ABSTRACT

Four classes of insecticide applied on seed were evaluated for managing high populations of the sugar beet wireworm, *Limonius californicus* (Coleoptera: Elateridae), in spring wheat in southern Alberta, Canada. Three separate field trials were conducted, and assessments made for stand protection, yield, and wireworm survival. Imidacloprid and thiamethoxam applied at 10–30 g AI and cyantraniliprole applied at 10–40 g AI provided initial stand protection, but did not protect seedlings until harvest and did not decrease wireworm populations. λ-cyhalothrin applied at 30 g AI provided stand protection that persisted until harvest, but yields were considerably lower than observed in fipronil treatments and there was little (23%) decrease in populations relative to controls. Fipronil applied at 0.6, 1.0, and 5.0 g AI, either singly or in blend with thiamethoxam at 10 g AI, provided stand protection until harvest and significantly reduced numbers of wireworms larger than 10 mm (range: 74–96%). Very low numbers of small (<11 mm) wireworms were observed in all trials. These results are compared to data from laboratory and field studies for this and other wireworm species. The relation between crop stand protection and wireworm mortality, the potential of insecticide blends, and the importance of seed type, wireworm species, and activity periods for managing wireworms with seed treatments are discussed.

Keywords: *Limonius californicus*, wireworm, pest management, thiamethoxam, fipronil, insecticide blend

INTRODUCTION

Wireworms have long been important insect pests in cereal, sugar beet, and potato production in southern Alberta (AB) (Strickland 1927). Historically, the main pest species were the prairie grain wireworm, *Selatosomus destructor* (Brown) and *Hypnoidus bicolor* (Esch.) (Strickland 1927; Arnason 1931). Recent surveys indicate *S. destructor* and *H. bicolor* remain the most commonly occurring elaterid pests in AB and Saskatchewan (SK), while the sugar beet wireworm, *Limonius californicus* (Mann.), is of more regional importance (van Herk and Vernon 2014). Described as an occasional pest new to AB in the 1950s (MacNay 1954), and historically found only in low numbers alongside *S. destructor* and *H. bicolor* (Doane 1977), *L. californicus* is currently the third most prevalent wireworm species in arable land in the Prairie Provinces (van Herk and Vernon 2014). In southern AB, where it is often the predominant species in continuously cropped cereals, high *L. californicus* populations can cause complete stand destruction of spring wheat, even if treated with insecticides (T.J. Labun, personal observation). The relatively recent emergence of this species as a pest in this region may stem from changes in cultivation practices, including the implementation of minimal tillage practices in recent decades which have increased soil moisture retention. *Limonius californicus* is

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known to prefer moist soils (e.g., irrigated land) and is typically not found on dry land (van Herk and Vernon 2014). Little else is known about the ecology and life history of this species, other than what was described by Stone (1941) for California, which suggests it is similar to the dusky wireworm, *Agriotes obscurus* L., and has a three- to five-year larval stage in the field.

In cereal production, wireworms have historically been managed by seed treatments, particularly chlorinated hydrocarbons (Toba *et al.* 1988; Grove *et al.* 2000). Treating seed with lindane decreased wireworm damage in cereal crops in the Canadian prairies by 90% and pest populations by 70% in the 1940s (Arnason and Fox 1948), and led to further decreases in damage between 1954 and 1961 (Burrage 1964). Similar results were obtained with other species, including *L. californicus*, in spring wheat in the Pacific Northwest (Toba *et al.* 1985, 1988). The effectiveness of lindane as a seed treatment stemmed from its ability to kill multiple pest species and all wireworm instars of these species, including neonates emerging from eggs laid after the seed is planted (Vernon *et al.* 2009). As a result of the latter property, wireworms would not repopulate fields to economic levels for several years after treatment, and farmers typically treated their cereal crops every 3–4 years (Arnason and Fox 1948). The reduction of wireworm populations achieved by planting lindane-treated seeds also protected high-value rotational crops such as sugarbeet, potato, and canola planted in subsequent years. Following lindane's de-registration (Canada in 2004; USA in 2006), there has been a gradual but continual increase in the incidence of wireworm damage in Canadian agricultural land. As a result, there is now a pressing need to identify and register new wireworm control measures for cereal crops. Such measures should be cost effective, pose negligible risk to humans and the environment, and offer similar efficacy to lindane by providing both stand and yield protection and reduction of wireworm populations (including controlling neonates) (Vernon *et al.* 2013a).

Initial results from laboratory and field evaluations of candidate insecticides to replace lindane as cereal seed treatments indicated neonicotinoid insecticides applied to wheat seed at 10–30 g AI/100 kg seed provide excellent stand and yield protection of spring wheat in the field in the presence of moderate to high populations of *A. obscurus*, but they did not decrease populations relative to control treatments (Vernon *et al.* 2009, 2013a). This disconnect between crop protection and lack of wireworm mortality was due to these insecticides inducing rapid and prolonged periods of morbidity, during which wireworms are unable to feed and after which they generally recover (Vernon *et al.* 2008, 2009). In contrast, the phenyl-pyrazole fipronil applied at 60 g AI/100 kg seed (a rate similar to that formerly registered for lindane) provided excellent stand and yield protection and eliminated *A. obscurus* populations (including neonate larvae) in the field (Vernon *et al.* 2009, 2013a). Laboratory studies indicated that dermal exposure of *A. obscurus* to fipronil causes rapid and irreversible morbidity, leading to complete mortality at higher rates. At low rates of fipronil exposure, wireworms showed no morbidity symptoms for several months, after which latent morbidity symptoms became apparent and mortality followed (Vernon *et al.* 2008). Exposing wireworms to wheat seed treated with low rates of fipronil permits them to feed normally until they succumb to latent mortality (Vernon *et al.* 2013a).

Based on these observations, we hypothesized that applying low rates of both thiamethoxam and fipronil to wheat seed would both provide stand and yield protection equivalent to lindane, and significantly reduce wireworm (including neonate) populations in the field (Vernon *et al.* 2013a). Specifically, thiamethoxam would provide early-season crop protection, while fipronil, even at very low doses, would cause late-season wireworm mortality. This approach would require low amounts of chemical, thereby reducing the environmental and human risk posed by these insecticides. Subsequent studies with *A. obscurus* demonstrated that blends of thiamethoxam at 10 g AI/100 kg seed and fipronil at 1 g AI/100 kg seed provided plant protection and wireworm control equivalent to lindane (Vernon *et al.* 2013a). Similarly, Morales-Rodriguez and Wanner

(2015) found that blends of these insecticides provide plant protection and reduce numbers of *L. californicus* and *H. bicolor*, but their field studies evaluated a single rate of these chemicals and under low pest pressure.

Here, we present results from three trials conducted in southern AB in fields with very high populations of *L. californicus* to determine the efficacy of these blends and other candidate insecticides. As wireworm species differ in their susceptibility to insecticides, the results presented here constitute an important extension to the efficacy data previously reported for other species.

MATERIALS AND METHODS

Plot layout and preparation. All three trials were conducted in 2012 near Granum, AB, on a commercial field (approx. 240 ha) that had been planted to barley, peas, and wheat in 2009, 2010, and 2011, respectively, and that had a recent history of wireworm damage. No insecticides had been applied to crops planted in this field since ca. 2000.

Experimental design. All trials were randomized complete block designs with four replicates. Each trial contained seed not treated with insecticide as a control treatment, and included a combined thiamethoxam (Cruiser 5FS at 10 g AI/100 kg seed) and fipronil (Regent 500FS at 1 g AI/100 kg seed) as a common insecticide seed treatment (hereafter referred to as 'Standard T+F Blend') to permit between-trial comparisons. Individual treatment plots in all trials consisted of seven 6.0-metre-long rows of wheat oriented due West to East, with 0.20 m spacing between treatment rows, 1.6 m between adjacent treatment plots, and 2.0 m between replicates.

Seed treatments. Seeds (hard red spring wheat: Syngenta, WR859CL) were treated with a Hege 11 liquid seed treatment applicator (Wintersteiger Inc., Salt Lake City, UT) by technicians at a Syngenta Crop Protection (Canada) seed treatment facility in Portage la Prairie, Manitoba, with the following insecticides:

Trial 1: Cyantraniliprole and λ-cyhalothrin: Cyantraniliprole (Fortenza 600FS) at 10, 20, 30, and 40 g AI/100 kg seed, λ-cyhalothrin (Demand 100CS) at 30 g AI, thiamethoxam (Cruiser 5FS) at 30 g AI, fipronil (Regent 500FS) at 5 g AI, and the Standard T+F Blend. All treatments also contained the fungicide Dividend XLRTA at 13 g AI (containing 3.21% difenoconazole and 0.27% mefenoxam).

Trial 2: Fipronil, alone and blended with thiamethoxam: Thiamethoxam (Cruiser 5FS) at 10 g AI/100 kg seed, fipronil (Regent 500FS) at 0.6, 1, and 5 g AI, and blends of thiamethoxam at 10 g AI + fipronil at 0.6, 1, and 5 g AI. All treatments also contained the fungicides Proseed at 2.5 g AI (containing 40.3% fludioxonil) and Vibrance XL at 17.5 g AI (containing 1.2% sedaxane, 5.9% difenoconazofe, and 1.5% metalaxyl-M).

Trial 3: Imidacloprid and thiamethoxam: Imidacloprid (Stress Shield 480SC) at 10, 20, and 30 g AI, thiamethoxam (Cruiser 5FS) at 10, 20, and 30 g AI, and Standard T+F Blend. The imidacloprid treatments also contained the fungicide Raxil MD at 3.5 g AI; all other treatments also contained the fungicides Proseed 480FS at 2.5 g and Vibrance XL at 17.5 g AI.

Planting*:* All plots were planted on 8 May 2012 with a seven-row double disc drill, no till planter (Fabro Enterprises Ltd., Swift Current, SK) directly into the wheat stubble from the previous year's crop. No tillage was done in the previous fall nor immediately prior to planting. Seeds were planted approx. 2.5 cm deep, at 285 seeds/m^2. As rows were spaced 20 cm apart, this seeding rate was equivalent to approx. 57 seeds per 1 m of row, or 100 kg seed/ha.

Stand assessment. Plant survival (hereafter "stand") was determined by counting the number of wheat seedlings alive in the central two-metre sections of the middle three rows of each plot at 14 and 29 days after planting (DAP) (22 May and 6 June, respectively) in all three trials, and measuring the plant reflective index (NDVI; Crop Circle ACS-430, Holland Scientific, Lincoln NE) at 21, 29, and 37 DAP (29 May, 6 and 14 June, respectively).

Plot maintenance: Plots were kept weed free by treating with glyphosate on 4 May prior to seeding, and no further weed control was deemed necessary. After harvest, the remaining wheat stubble was left intact over winter to prevent disturbance of surviving wireworm populations, which were assessed by trapping the following spring.

Harvest. All trials were harvested on 28 August 2012 (112 DAP) using a small plot combine (Wintersteiger Inc., Salt Lake City, UT) that calculated both the moisture percentage in the seed and per hectare yield. Some plots (e.g., neonicotinoid treatments in Trial 3) were not harvested due to the lack of surviving plants.

Wireworm trapping. To determine the longer-term effects of the various treatments on wireworm mortality, wireworms were sampled in the spring of the following year using a bait-trapping procedure similar to that described in Vernon *et al.* (2009). Bait traps were installed in the plots (three per plot) on 1–2 May, 2013, and removed on 13 May. Bait trap locations were spaced 1 m apart along the middle of each plot, so that the traps were 2, 3, and 4 m from the front and 75 cm from the outer rows of each plot. Each bait trap consisted of a 450-ml plastic flower pot filled with coarse-grade vermiculite and 100 ml untreated hard red spring wheat placed in a layer in the middle of the pot. Traps were soaked to run-off with lukewarm water twice several hours before placement in circular holes (10 cm diameter, 15 cm deep) cored into the ground. Soil was carefully and consistently packed around and on top of the bait traps, and a 20-centimetre-diameter inverted tray placed 5 cm above the trap and level with the ground. To reduce variability in data, considerable effort was taken to ensure each trap was prepared and installed identically. After removal, bait traps were immediately transported to the Agassiz Research and Development Centre (AAFC, Agassiz, BC), where they were placed in Tullgren funnels on 15 May for 2 weeks to extract wireworms. Extracted wireworms were counted, measured to the nearest millimetre, and identified to species. As wireworms shrink when they desiccate after extraction, 200 living *L. californicus* larvae were individually placed directly under the funnel heat source (25W incandescent light bulbs) for 48 h, and measured and weighed to 0.1 mg (Sartorius CP64 analytic balance; Sartorius AG, Goettingen, Germany) both before and after desiccation. Simple linear regression of desiccated to living wireworm length yielded the relation, living length = (desiccated length + 0.5391) / 0.6655; $R^2 = 0.81$, which was subsequently used to convert the lengths of desiccated wireworms to the corresponding size of living ones. For analyses, larval lengths were combined in three millimetre categories, since binning into two millimetre categories or showing each size separately would produce artifacts due to sizes calculated from desiccated lengths being rounded to the nearest 1 mm, which causes underestimations of the number that were 6, 9, 12 mm, etc. long. Wireworms were considered small, or neonate, if equal or less than 10 mm long, and large (or resident) if greater than 10 mm.

Statistical Analysis. All data analyses were conducted using SAS (SAS 9.2, SAS Institute, Cary, NC). Treatment means were compared by ANOVA (Proc GLM), followed by mean separation with Tukey's standardized range honestly significant difference (HSD) test at $\alpha = 0.05$. Where data could not be easily normalized using a power transformation (Trial 3, reflective index and yield only), the Kruskal–Wallis test (Proc NPAR1WAY) was used, after which normalized rank values were assigned to treatments (Proc RANK) and the standard ANOVA and the Tukey procedures performed on the rank values. The relationship between the amount of stand reflectivity and plant stand counts recorded on the same day was analyzed with linear regression (Proc GLM). Count data were analyzed with chi-square tests (Proc FREQ).

RESULTS

Wireworm sampling

Post-treatment bait-trapping results confirmed the trial areas had very high wireworm populations, with 403 larvae collected from the combined control treatments in the three

trials (12 plots, 36 traps), and 2,234 from the combined treated plots (88 plots). Of the latter, only 190 wireworms were in plots with seeds treated with fipronil alone or in blend with another insecticide (36 plots). Similar numbers were found in the control plots of all three trials (range of means: 8.8–13.4/trap, Tables 1–3), suggesting a fairly homogeneous population in the study area. Wireworm populations were predominantly *L. californicus* (97.3%), with very low numbers of *H. bicolor* (2.0%), *S. destructor* (0.7%), and *Aeolus mellillus* (Say) (<0.1%) — the latter species are included in the totals presented in Tables 1–3. To compare the age structures of wireworm populations retrieved from the various treatments, the distribution of larval lengths (range: 3–28 mm) were compared for wireworms retrieved from control plots, plots seeded to treatments containing fipronil, and plots seeded to treatments containing other insecticides (Fig. 1). Chi-square analyses indicated significant differences in population structures (i.e., in the relative number in each of the size classes), both between control and fipronil treatments (Chi=1089.3, df=7, $P<0.0001$) and between control and other treatments (Chi=144.9, df=7, $P<0.0001$). Comparison of the age structures (Fig. 1A–C) indicates control treatments had significantly lower numbers of small (3–10 mm) wireworms per plot (1.08) than fipronil (1.94) and non-fipronil (2.92) insecticide treatments (Chi=104.3, df=1, $P<0.0001$; Chi=8.85, df=1, $P=0.0016$; respectively). In contrast, the control and non-fipronil treatments had a similar number of large (>10 mm) wireworms per trap (32.5 and 36.4, respectively per plot), while very low numbers (3.3 per plot) were retrieved from treatments containing fipronil (Fig. 1A–C).

Relation between plant reflectivity and plant stand

A direct, highly significant relationship was observed between plant reflectivity index (RI) and plant stand when the two were measured on the same day (29 DAP). This was true for trials with cyantraniliprole (t=7.95, df=1,34, $P<0.0001$, $R^2=0.64$), fipronil (t=11.17, df=1,30, $P<0.0001$, $R^2 = 0.80$), and imidacloprid and thiamethoxam (t =7.06, df=1,30, $P<0.0001$, $R^2=0.61$), and indicates that plant RI is an acceptable metric for assessing plant stand (e.g., at 37 DAP, when individual plant counts were not conducted).

Trial 1: Cyantraniliprole and λ-cyhalothrin

Stand protection and yield

Greatest stand protection was provided by fipronil at 5 g AI and Standard T+F Blend treatments. These treatments had higher stand counts than the control at 14 DAP (1.55x) and 29 DAP (6.03x, 5.61x, respectively). However, RI readings at 37 DAP indicate better stand protection in fipronil (5 g AI) than the Standard T+F Blend (2.08x vs 1.77x control, respectively), which resulted in higher yields at harvest (respectively, 18.3 vs 11.5x the control; Table 1). Thiamethoxam applied at 30 g AI provided good initial plant protection (respectively, 1.54x and 3.14x control at 14 and 29 DAP), but the RI at 37 DAP and yield at harvest were similar to control and significantly less than fipronil (5 g AI) and Standard T+F Blend treatments. Similarly, λ-cyhalothrin at 30 g AI provided stand protection (respectively, 1.80x and 4.60x control at 14 and 29 DAP) that resulted in similar yield to the Standard T+F Blend, but yield was significantly lower than observed for fipronil at 5 g AI (Table 1).

Cyantraniliprole applied at 10–40 g AI provided stand protection equivalent to or greater than thiamethoxam at 30 g AI (i.e., 1.60-1.80x control at 14 DAP, 3.02-3.85x control at 29 DAP), which resulted in numerically higher yields (1.73-2.56x thiamethoxam). Stand protection with cyantraniliprole was equivalent to that provided by λ-cyhalothrin and fipronil (5 g AI) at 14 DAP, but this had diminished by 29 DAP (0.50-0.64x fipronil), and the RI at 37 DAP and yields at harvest were significantly lower than fipronil (5 g AI) (Table 1). There were no significant differences in stand protection or yield between rates of cyantraniliprole (Table 1).

Table 1

Plant stand, crop yield, and wireworm survival in plots treated with cyantraniliprole and λ-cyhalothrin. Shown are mean (SE) values, based on four replicates (Rep). Wireworm numbers are calculated per plot (i.e., three bait traps combined). Plant stand (number of plants per 6.0-m row) and plant reflective index were measured at 14, 29, and 37 days after planting (DAP). Wireworms (wws) were considered 'large' if >10mm and 'small' if ≤10mm long (see text for explanation). Numbers followed by the same letter in a column are not significantly different at $\alpha = 0.05$.

Treatment *	Rate (g AI/ 100kg seed)	Plant stand: 14 DAP	Plant stand: 29 DAP	Reflective Index: 37 DAP	Yield (kg/ha) at harvest	Small wws	Large wws	All wws
Control	13	79.8 (5.7) B	25.3 (4.6) C	0.13 (0.008) D	206.8 (206.8) C	1.3 (0.5) AB	39.0 (5.5) A	40.3 (5.8) A
Cruiser 5FS	30	122.5 (12.7) AB	86.3 (28.2) ABC	0.15 (0.013) CD	373.2 (171.3) C	0.5 (0.5) B	32.8 (2.8) A	33.3 (3.4) A
Regent 500FS	5	124.0 (10.4) AB	152.5 (10.9) A	0.27 (0.021) A	3675.3 (295.8) A	0.8 (0.5) AB	2.2 (1.1) B	3.0 (1.5) B
Cruiser 5FS + Regent 500FS	10+1	123.5 (16.0) AB	142.0 (13.0) AB	0.23 (0.023) AB	2305.0 (177.9) B	1.3 (0.5) AB	8.2 (2.1) B	9.5 (1.9) B
Demand 10CS	30	143.3 (16.2) A	116.3 (13.3) AB	0.21 (0.007) BC	2342.6 (271.9) AB	1.0 (1.0) AB	30.0 (4.9) A	31.0 (5.7) A
Fortenza 600FS	10	143.8 (15.1) A	87.5 (19.0) ABC	0.16 (0.017) CD	955.0 (249.1) BC	1.5 (0.9) AB	46.5 (4.9) A	48.0 (5.5) A
Fortenza 600FS	20	135.8 (10.7) A	76.3 (14.0) BC	0.16 (0.009) CD	643.9 (226.7) C	2.3 (0.9) AB	48.5 (7.0) A	50.8 (6.9) A
Fortenza 600FS	30	127.8 (13.6) AB	97.3 (19.6) AB	0.17 (0.027) BCD	912.9 (445.9) BC	3.5 (0.9) A	44.5 (6.5) A	48.0 (7.3) A
Fortenza 600FS	40	130.8 (7.3) A	90.8 (14.3) ABC	0.15 (0.005) CD	955.0 (210.2) BC	3.0 (0.7) AB	39.3 (4.8) A	42.3 (5.4) A
Trt df=8,23		F=3.14, P=0.015	F=6.90, P=0.0001	F=12.11, P<0.0001	F=17.91, P<0.0001	F=2.87, P=0.02	F=16.46, P<0.0001	F=15.78, P<0.0001
Rep df=3,23		F=4.31, P=0.015	F=4.05, P=0.019	F=6.35, P=0.0027	F=0.51, P=0.68	F=4.60, P=0.011	F=4.53, P=0.012	F=5.11, P=0.007

All treatments contained the fungicide Dividend XLRTA at 13 g AI

Table 2

Plant stand, crop yield, and wireworm survival in plots treated with fipronil alone or in blend with thiamethoxam. Shown are mean (SE) values, based on four replicates (Rep). Wireworm numbers are calculated per plot (i.e., three bait traps combined). Plant stand (number of plants per 6.0-m row) and plant reflective index were measured at 14, 29, and 37 days after planting (DAP). Wireworms (wws) were considered 'large' if >10mm and 'small' if ≤10mm long (see text for explanation). Numbers followed by the same letter in a column are not significantly different at $\alpha = 0.05$

Treatment *	Rate (g AI/ 100kg seed)	Plant stand: 14 DAP	Plant stand: 29 DAP	Reflective Index: 37 DAP	Yield (kg/ha) at harvest	Small wws	Large wws	All wws
Control		83.5 (10.1) B	43.5 (15.6) C	0.13 (0.009) C	210.2 (205.7) D	1.0 (0.7) A	25.3 (3.8) A	26.3 (3.5) A
Cruiser 5FS	10	97.0 (9.3) AB	71.8 (6.1) BC	0.13 (0.006) C	432.1 (143.9) D	2.3 (0.3) A	30.0 (2.3) A	32.3 (2.4) A
Regent 500FS	0.6	104.3 (3.7) AB	116.3 (5.1) AB	0.19 (0.019) ABC	3024.6 (152.1) BC	1.3 (0.5) A	4.0 (1.6) B	5.3 (1.9) B
Regent 500FS	1	132.8 (8.9) AB	145.3 (10.0) A	0.20 (0.009) ABC	2891.8 (333.6) BC	3.3 (1.3) A	5.8 (1.9) B	9.0 (2.9) B
Regent 500FS	5	135.0 (18.0) AB	160.5 (10.9) A	0.26 (0.010) A	3767.7 (183.9) AB	0.5 (0.5) A	0.8 (0.5) B	1.3 (0.5) B
Cruiser 5FS + Regent 500FS	10+0.6	118.5 (16.3) AB	115.5 (13.7) AB	0.17 (0.012) BC	2797.6 (114.4) C	3.0 (1.1) A	6.5 (1.4) B	9.5 (2.2) B
Cruiser 5FS + Regent 500FS	10+1	117.5 (15.0) AB	135.0 (19.4) A	0.21 (0.026) ABC	3542.4 (249.0) ABC	0.3 (0.3) A	2.0 (1.2) B	2.3 (1.4) B
Cruiser 5FS + Regent 500FS	10+5	142.8 (16.1) A	164.0 (21.8) A	0.24 (0.034) AB	4016.6 (216.7) A	1.5 (0.6) A	0.8 (0.8) B	2.3 (1.1) B
Trt df=7,21		F=2.95, P=0.026	F=10.32, P<0.0001	F=7.17, P=0.0002	F=60.34, P<0.0001	F=2.51, P=0.048	F=34.19, P<0.0001	F=27.32 P<0.0001
Rep df=3,21		F=2.58, P=0.08	F=1.94, P=0.15	F=2.38, P=0.10	F=2.97, P=0.06	F=1.99, P=0.15	F=0.63, P=0.61	F=0.50, P=0.68

* All treatments contained the fungicides Proseed at 2.5 g AI and Vibrance XL at 17.5 g AI

Table 3

Plant stand, crop yield, and wireworm survival in plots treated with imidacloprid and thiamethoxam. Shown are mean (SE) values, based on four replicates (Rep). Wireworm numbers are calculated per plot (i.e., three bait traps combined). Plant stand (number of plants per 6.0-m row) and plant reflective index were measured at 14, 29, and 37 days after planting (DAP). Wireworms (wws) were considered 'large' if >10mm and 'small' if ≤10mm long (see text for explanation). Numbers followed by the same letter in a column are not significantly different at $\alpha = 0.05$.

Treatment *	Rate (g AI/ 100kg seed)	Plant stand: 14 DAP	Plant stand: 29 DAP	Reflective Index: 37 DAP **	Yield (kg/ha) at harvest **	Small wws	Large wws	All wws
Control		88.8 (8.7) B	29.5 (5.6) C	0.12 (0.003) A	0 (0) A	0.8 (0.8) A	33.5 (6.5) A	34.3 (6.9) A
Stress Shield 480SC	10	118.0 (9.1) AB	72.0 (12.2) BC	0.13 (0.009) ABC	0 (0) A	1.8 (1.4) A	38.5 (6.8) A	40.3 (8.2) A
Stress Shield 480SC	20	141.5 (9.6) A	91.8 (13.0) ABC	0.13 (0.009) ABC	0 (0) A	3.3 (1.7) A	25.0 (3.7) AB	28.3 (5.2) AB
Stress Shield 480SC	30	151.5 (17.9) A	101.8 (18.8) AB	0.14 (0.009) BC	0 (0) A	3.3 (1.4) A	38.5 (2.6) A	41.8 (1.9) A
Cruiser 5FS	10	109.5 (11.0) AB	38.5 (7.5) C	0.12 (0.002) AB	0 (0) A	2.0 (0.8) A	26.5 (8.7) AB	28.5 (9.0) AB
Cruiser 5FS	20	133.5 (14.4) AB	66.3 (4.9) BC	0.13 (0.003) ABC	0 (0) A	1.5 (0.3) A	39.0 (5.6) A	40.5 (5.4) A
Cruiser 5FS	30	137.5 (14.5) A	76.0 (13.4) BC	0.13 (0.005) ABC	0 (0) A	2.3 (0.9) A	44.0 (5.5) A	46.3 (5.3) A
Cruiser 5FS + Regent 500FS	10+1	140.5 (10.2) A	144.0 (18.8) A	0.18 (0.019) C	2824.5 (326.4) B	4.0 (2.5) A	1.5 (0.9) B	5.5 (2.4) B
Trt df=7,21		F=4.41, P=0.004	F=7.57, P=0.0001	F=4.89, P=0.0021	F=95.67, P<0.0001	F=0.60, P=0.75	F=5.86, P=0.0007	F=4.60, P=0.003
Rep df=3,21		F=5.52, P=0.006	F=0.59, P=0.63	F=2.65, P=0.08	F=1.00, P=0.41	F=0.57, P=0.64	F=0.98, P=0.42	F=1.00, P=0.41

* Stress Shield 480SC treatments contained the fungicide Raxil MD at 3.5 g AI; all other treatments contained the fungicides Proseed at 2.5 g AI and Vibrance XL at 17.5 g AI

** ANOVA and mean separation conducted on normalized ranks. The non-parametric Kruskal–Wallis test was used to conduct initial analyses — Reflective Index: 37 DAP: H=15.87, df=1, P=0.0266; Yield (kg/ha) at harvest: H=30.83, df=1, P<0.0001.

Wireworm survivorship

Significantly fewer large (>10 mm) wireworms were collected in bait traps in both the fipronil (0.06x control) and Standard T+F Blend (0.21x control) treatments (Table 1), indicating high mortality in these treatments. In contrast, there were no significant reductions in large wireworms caught in the thiamethoxam (30 g AI), λ-cyhalothrin (30 g AI), and cyantraniliprole treatments relative to the control treatment (Table 1). Relatively few small (neonate) wireworms were collected in all insecticide treatments, and this was similar to numbers taken in the control treatment (Table 1).

Trial 2: Fipronil, alone and blended with thiamethoxam

Stand protection and yield

Higher stand protection was observed in fipronil (0.6, 1.0, and 5.0 g AI) treatments relative to the untreated control (range: 1.25–1.62x stand at 14 DAP, 2.67–3.69x stand at 29 DAP) and the thiamethoxam (10 g AI) treatments (1.08–1.39x stand at 14 DAP, 1.62–2.24x stand at 29 DAP). Stand protection increased with the rate of fipronil applied. As in the other trials, thiamethoxam failed to provide lasting plant protection, leading to very low yields at harvest (Table 2). In contrast, all rates of fipronil provided significantly higher yields than either the thiamethoxam or untreated control treatments (13.76–17.92x control; Table 2). No significant differences in yield were observed between the fipronil rates. Combining thiamethoxam at 10 g AI with fipronil at 0.6, 1.0, or 5 g AI provided similar stand protection than the fipronil treatments alone at the same rates, and did not significantly increase yields (13.31–19.11x control; Table 2).

Wireworm survivorship

Populations of large wireworms were significantly reduced in the fipronil (0.6, 1.0, and 5.0 g AI) (range: 0.03–0.23x control) and combined thiamethoxam (10 g AI) and fipronil (0.6, 1.0, and 5.0 g AI) treatments (0.03–0.26x control) (Table 2). Mortality was highest in treatments with the 5 g AI rate of fipronil. Although not statistically significant, there was notably higher mortality in the Standard T+F Blend than in the treatment with fipronil at 1 g AI alone (Table 2). In contrast, more (1.19x control) large wireworms were collected from the thiamethoxam than the control treatment (Table 2). Low and similar numbers of neonate wireworms were collected from all treatments.

Trial 3: Imidacloprid and thiamethoxam

Stand protection and yield

Both imidacloprid (10, 20, and 30 g AI) and thiamethoxam (10, 20, and 30 g AI) provided initial stand protection (1.33–1.71x, 1.23–1.55x control at 14 DAP, respectively; 2.44–3.45x, 1.31–2.58x control at 29 DAP). For each rate tested, imidacloprid provided numerically greater protection than thiamethoxam, with protection increasing with rate for both chemicals (Table 3). Stand protection disappeared after 37 DAP, leading to complete destruction of the plots and no harvestable plants.

Good initial plant protection was observed in the Standard T+F Blend (1.58x and 4.88x control at 14 and 29 DAP, respectively). The effect of fipronil in the Standard T+F Blend was evident when compared to thiamethoxam applied alone at 10 g AI (1.28x and 3.74x thiamethoxam at 14 and 29 DAP, respectively). Plant stand protection in the Standard T+F Blend persisted throughout the season, and this was the only treatment with harvestable plants. Yields at harvest were similar to that observed for the same treatment evaluated in the other two trials (respectively, 2305, 3542, and 2824 kg/ha, Tables 1–3).

Wireworm survivorship

Populations of large wireworms were not reduced in any of the imidacloprid or thiamethoxam treatments (range: 0.75–1.15x, 0.79–1.31x control, respectively), and highest numbers were collected from plots seeded to the highest rates of these chemicals (Table 3). In contrast, very low numbers of large wireworms (0.04x control) were collected from the Standard T+F Blend treatment, indicating high mortality. Low and similar numbers of neonate larvae were collected from all treatments (Table 3).

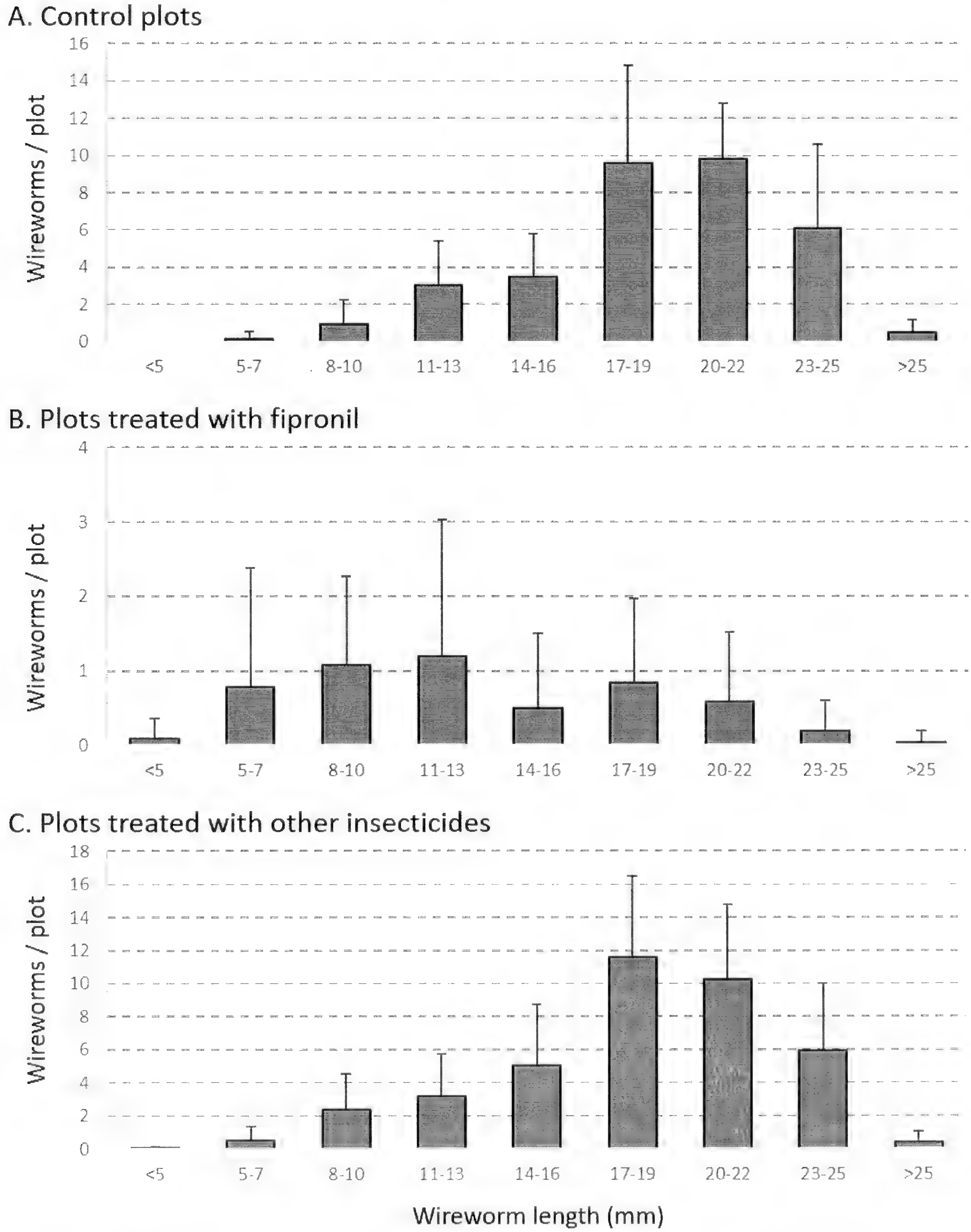

Figure 1. Size distribution of wireworms (predominantly *Limonius californicus*) collected from three insecticide efficacy trials conducted in Claresholm, Alberta. Mean (SD) number of wireworms retrieved from bait traps placed in control plots (**A.**, N = 12 plots), in plots treated with fipronil alone or in blend with another insecticide (**B.**, N = 36), and in plots treated with an insecticide other than fipronil (**C.**, N = 52). Note the differences in vertical axes between **B** and **A, C.**

DISCUSSION

Neonate versus resident wireworm mortality

The number of small (neonate) wireworms that would have been produced during this study was low (approx. 10%) in all treatments relative to the number of large (resident) wireworms that would have been present at the time of planting. This is in contrast to field studies with *A. obscurus* in which higher numbers of neonates were trapped in control plots and plots treated with neonicotinoids relative to fipronil-containing plots (cf. Vernon *et al.* 2009). There are a number of possible reasons for the differences in neonate catches between the previous and current studies. In the current study, plant stand in some (e.g., control, neonicotinoids; Tables 1–3) treatments was poor to non-existent, which would have reduced oviposition and food availability relative to treatments with higher stands (e.g., fipronil-containing treatments). This is partially substantiated by the cyantraniliprole treatments in Trial 1, where stand and yield were higher than in the control treatment, and neonate numbers were numerically higher (4.0–4.8 per plot) than in the other treatments (1.5–2.7 per plot) (Table 1). This also suggests cyantraniliprole may not be lethal to neonate wireworms.

In plots containing fipronil, which had excellent stand protection, low neonate numbers were likely due to the residual and toxic effect of this chemical. Numbers of resident wireworms were also very low in these treatments, and fipronil has previously been shown to be highly toxic to both resident and neonate *A. obscurus* (Vernon *et al.* 2009, 2013a, 2016). The effect of the pyrethroid, λ-cyhalothrin, in reducing neonate populations in the current study is more difficult to ascertain. Because stand protection and yield were similar to the Standard T+F Blend, the reduction in resident populations was not significantly different from thiamethoxam or the control (Table 1), and the neonate numbers were low, it appears that λ-cyhalothrin is persistent and toxic to this stage and/or that the presence of this insecticide in plots reduced egg laying due to repulsion of female beetles. We have previously shown that residues of another pyrethroid, bifenthrin, are repulsive to *A. obscurus* larvae >200 d after an in-furrow application to soil in potatoes (van Herk *et al.* 2013). While the overall low number of neonates in this study might be attributed to low click beetle emergence and egg-laying, this typically occurs in fields treated with an insecticide (e.g., that induces prolonged morbidity and prevents late-instar larvae from feeding sufficiently to pupate in the fall), whereas no insecticides had been applied to the study field since approx. 2000 (T.J. Labun, unpublished data).

It is interesting that the lack of food in certain plots did not appear to affect the survival and retention of resident wireworms, with high numbers of larvae trapped from plots with little or no plant survival (e.g., neonicotinoid treatments, Table 3). This supports the concern that later instars of some pest species can survive with minimal food for prolonged periods of time (Vernon and van Herk 2013). Also worth noting is that none of the fungicide treatments used in these trials appeared to negatively affect wireworm populations. This is consistent with results from lab and field studies with both *A. obscurus* and *L. canus* LeC. (Vernon *et al.* 2009, 2013a; van Herk *et al.* 2008, 2015).

Crop protection vs. wireworm mortality, and benefits of blended treatments

The above results underscore the importance of evaluating wireworm mortality (inferred here from the difference in wireworm numbers collected from treatment vs control plots) in field efficacy studies. While wireworm mortality could be deduced from crop protection in earlier insecticide efficacy studies with OP and OC insecticides, this is usually not possible with newer chemistries (Vernon *et al.* 2009), as exposure to neonicotinoid insecticides generally induces prolonged, reversible morbidity during which time wireworms are unable to feed (Vernon *et al.* 2008). Hence, these insecticides may protect plants from feeding damage without decreasing wireworm populations (Vernon *et al.* 2009, 2013a). A similar result was seen in efficacy studies with potatoes, where neonicotinoid treatments applied at planting reduced feeding damage to daughter

tubers without decreasing wireworm numbers (Vernon *et al.* 2013b). Pyrethroid insecticides also protect wheat and potatoes from wireworm feeding damage without reducing populations, but here the mechanism is mainly repellency (van Herk *et al.* 2008, 2015). Conversely, exposure to an insecticide that induces morbidity and mortality latently can result in wireworm population reductions without providing adequate stand protection (Vernon *et al.* 2013a).

Contrary to results with *A. obscurus* in BC, high rates of imidacloprid and thiamethoxam failed to protect wheat seedlings from *L. californicus* past 29 DAP in these trials. This could result from differences in insecticide susceptibility between species or from the very high wireworm populations in the field. In southern Alberta, high populations of *L. californicus* can cause complete crop destruction in fields of spring wheat treated with a high (39 g AI) rate of thiamethoxam (T.J. Labun, personal observation). The observed failure of high rates of these commonly used insecticides to reduce populations of *L. californicus* is similar to findings by Esser *et al.* (2015) with *L. californicus* and *L. infuscatus* Mots., and likely explains why damage in wheat from these species is increasing in severity and frequency across the region.

Both cyantraniliprole and λ-cyhalothrin provided greater protection at the rates tested than either imidacloprid or thiamethoxam, although this was likely through different mechanisms. While λ-cyhalothrin and other pyrethroids (e.g., tefluthrin, bifenthrin) induce repellency and thereby reduce feeding (van Herk *et al.* 2008, 2015), cyantraniliprole is not repulsive and likely induces morbidity after feeding (van Herk *et al.* 2015). Considering the high wireworm populations in these trials, the partial plant protection observed is encouraging, and cyantraniliprole may be a potential candidate for blending with low rates of a lethal insecticide. It should be noted that at the rates tested, cyantraniliprole and λ-cyhalothrin by themselves did not cause significant wireworm mortality in either this study or in previous work with *A. obscurus* (Vernon *et al.* 2013b; van Herk *et al.* 2015).

Combining a non-lethal insecticide that rapidly induces morbidity with a low rate of a chemical that causes mortality latently can provide both stand protection and long-term population reductions in the field (Vernon *et al.* 2013a). Since wireworms live for up to 4–5 years in the soil, one application with an insecticide lethal to all wireworm stages can remove the economic threat of wireworms for three or more years. This blended treatment concept was evaluated in numerous lab and field studies with *A. obscurus, A. sputator,* and *L. canus,* which demonstrated that combinations of thiamethoxam at 5 or 10 g AI with fipronil at rates as low as 1 g AI will provide both acceptable crop protection and high neonate and resident wireworm mortality for these species (Vernon *et al.* 2009, 2013a). These results provided the basis for the current study with *L. californicus* and allowed the concept to be extended to using insecticide-blended wheat seed as an in-furrow treatment that both protects potato tubers from damage and reduces wireworm populations (Vernon *et al.* 2016).

In the work reported here, both the fipronil and various thiamethoxam + fipronil blend treatments provided significant stand protection and reduction in populations of resident wireworms, relative to the untreated control and all other treatments tested. Of note is that, in Trial 2, combining thiamethoxam at 10 g AI with fipronil at 0.6, 1.0, and 5.0 g AI did not improve stand protection and yield, nor increase resident wireworm mortality relative to the corresponding fipronil treatments. This suggests that *L. californicus* may respond differently to neonicotinoid and fipronil insecticide blends than *A. obscurus*, where the presence of thiamethoxam considerably improved stand and yield (Vernon *et al.* 2013a). Also of note is that stand, yield, and mortality were notably higher at the 5.0 g than 1.0 g and 0.6 g AI rates of fipronil. Similarly, in Trial 1, fipronil at 5 g AI provided 1.6x greater yield and 3.6x higher mortality than the Standard T+F Blend. This suggests that where fipronil is used alone as a seed treatment to control high populations of *L. californicus*, it should be applied at a rate higher than 1 g AI, and that (unlike for *A.*

obscurus) there is no additional benefit from combining fipronil with a neonicotinoid such as thiamethoxam.

Neonicotinoid and fipronil insecticide blends on wheat seed have been evaluated for wireworm management elsewhere. Morales-Rodriguez and Wanner (2015) observed high (>70%) mortality in *L. californicus* and *H. bicolor* exposed in laboratory assays to wheat seed treated with fipronil at 1 and 5 g AI/100 kg seed but low mortality (<30%) if exposed to thiamethoxam at 39 g AI. In field trials, seed treated with thiamethoxam at 39 g AI provided plant protection but resulted in higher wireworm populations than control plots, while seed treated with both thiamethoxam at 39 g AI and fipronil at 5 g AI significantly reduced populations. Combining thiamethoxam at 39 g AI with fipronil at 1 g AI/100 kg seed caused less mortality in lab studies than either insecticide alone, and we suggest that the high rate of thiamethoxam in this blend may have induced morbidity before sufficient fipronil was ingested. Higher rates of thiamethoxam decrease the duration of feeding in *L. canus* (van Herk *et al.* 2008), and in lab studies mortality is greater when wireworms are exposed to fipronil at 1 g AI alone than in combination with thiamethoxam at 10 g AI (van Herk *et al.* 2015). However, when larvae were exposed to a blend of thiamethoxam at 10 g AI and a higher rate of fipronil (e.g., 5 g AI), enough of the latter chemical was ingested to cause high mortality (van Herk *et al.* 2015). Under field conditions, high mortality of *A. obscurus* was observed with blends of thiamethoxam at 5 or 10 g AI and fipronil at both 1 and 5 g AI (Vernon *et al.* 2013b), likely because of longer exposure to the seeds than in laboratory studies and because other factors (i.e., desiccation, predation on moribund wireworms) contribute to mortality in the field (Vernon *et al.* 2009).

Potential of seed treatments for controlling wireworms in cereals

In a recent review of insecticides for controlling wireworms in cereals, it was observed that, in general, the most effective chemistries appear to be those that target GABA-gated chloride channels (e.g., fipronil, lindane) (van Herk *et al.* 2015). As noted by Lange *et al.* (1949), the efficacy of seed treatments also depends on "the species of wireworms involved, wireworm activity at the time the seed is planted, the proportion of the population attracted to the seed, the type of seed, and the time of planting." Some of these observations are briefly considered here.

Time of planting and wireworm activity

Seed treatments are most likely to be effective when seed is planted shortly before larvae become active (Vernon and van Herk 2013). Many pest wireworm species have two main periods of feeding activity (spring and fall), between which they burrow downwards to avoid desiccation (Traugott *et al.* 2015). Planting seed treated with a non-residual insecticide after wireworms have fed would therefore reduce exposure and resultant mortality. This would be a concern where cropping practices (e.g., continuous cropping, minimal tillage) provide alternative food sources before or after the seeds are planted (e.g., roots and decaying plant matter from the previous year's crop). Under these conditions, wireworms would presumably feed less on the treated seeds, if at all, and therefore ingest less insecticide (Vernon *et al.* 2013b). Early season planting, before wireworms become active in the spring, may not be feasible, as wireworms can cause considerable feeding damage even at low soil temperatures (van Herk and Vernon 2013).

Determining when wireworms become active in the spring has been the focus of considerable research (reviewed in Traugott *et al.* 2015 and Vernon and van Herk 2013), and the high mortality observed in the fipronil treatments reported here suggests the spring activity period of *L. californicus* coincides with spring wheat planting in southern Alberta.

Differences between species

Insecticide seed treatment efficacy may vary between wireworm species due to differences in species phenology (e.g., when they begin to feed) and different susceptibilities to insecticides (Vernon *et al.* 2008). Lange *et al.* (1949) noted that *L. canus* is more susceptible to lindane than *L. californicus,* possibly because of differences

in the activity levels of these species. In eastern Washington State, repeated exposure to thiamethoxam-treated spring wheat resulted in no observed changes in populations of *L. californicus*, whereas at a nearby site it appeared to reduce *L. infuscatus* populations (Esser *et al.* 2015; Milosavljevic *et al.* 2016). Hence, it is critically important to know what species are present in the field before applying a management approach, particularly as pest species frequently co-occur.

Differences between cereals

In laboratory studies, Edwards and Evans (1950) observed no difference in wheat and oat (*Avena sativa* L.) seedling survival when exposed to *Corymbites cupreus* Fabr., *Agriotes* spp., or *Athous (=Hemicrepidius) niger* L. larvae, but slightly higher survival of barley (*Hordeum vulgare* L.) than wheat and oat seedlings exposed to *Agriotes* spp. and *C. cupreus*. In contrast, recent work suggests both oat and barley seedlings may be less susceptible to *L. infuscatus* and *L. californicus* feeding (respectively) than wheat (Higginbotham *et al.* 2014, Rashed *et al.* 2017). Recent field studies in Alberta suggest insecticides (e.g., fipronil) applied on barley cause lower mortality in *L. californicus* than when applied to spring wheat seed (van Herk *et al.*, unpublished data). This may be due to the barley seed hull absorbing some of the seed dressing, or to the susceptibility of the seed itself to wireworm feeding (cf. Higginbotham *et al.* 2014). While more data is required to determine if these results are real or result from the usual sources of variability that plague wireworm field studies (e.g., patchy distributions in the field), insecticides used as seed treatments may need to be applied at higher rates on barley than wheat to achieve the same level of population reduction, but at lower rates to achieve the same level of stand protection.

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SCIENTIFIC NOTE

A pheromone-baited pitfall trap for monitoring *Agriotes* spp. click beetles (Coleoptera: Elateridae) and other soil-surface insects

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Pheromone traps have been developed specifically for the survey, research and management of click beetles (Coleoptera: Elateridae) in temperate North America (NA), Europe and Asia (Ritter and Richter 2013; Vernon and van Herk 2013; Traugott *et al.* 2015). These include: 'Estron Traps' for survey of *Agriotes* species in the former USSR (Oleshchenko *et al.* 1987); 'Yatlor Traps' for survey and scientific study of *Agriotes* in Europe (Furlan *et al.* 2001), and; 'Vernon Beetle Traps' for survey and integrated pest management (IPM) of invasive *Agriotes* in NA, i.e., *A. obscurus* (AO), *A. lineatus* (AL) and *A. sputator* (AS; Vernon 2004). Although effective, these traps are no longer available commercially, although the Yatlor Trap has been re-designed as a funnel trap to better intercept various flying *Agriotes* in Europe (Csalomon, Budapest, Hungary). The loss of the Vernon Beetle Trap (VBT) and customized lures for AO, AL and AS [formerly produced by Contech Enterprises Inc., Delta, British Columbia (BC), Canada] necessitated the development of a new trap for use in *Agriotes* IPM program development in Canada. Based on the authors' experience with earlier *Agriotes* traps, the new trap was designed to: provide trapping efficacy comparable to the VBT; reduce the time required for assembly, installation and inspection; exclude insectivorous vertebrates and water, and; be consistent, reliable, inexpensive, small, easy to transport, and durable.

The new trap, named the Vernon Pitfall Trap® (VPT) (Fig. 1), is constructed of durable polypropylene, and is formed from three custom injection molds (Exact Molds Ltd, Abbotsford, BC). Two essential components are an in-ground pitfall chamber for specimen collection (Fig. 1A) and a protective cover containing a pheromone-bait holder and vertebrate-exclusion cage (Fig. 1B). The pitfall chamber forms a tapered cup that is 10 cm high from base to apex of the trap, with a 5.8-cm-diameter base (inside diameter, ID) and a 9-cm-diameter opening (ID) (Fig. 1A). The inside of the cup, three centimetres from the apex, is molded to receive a commercially available specimen cup (specifically, Fisherbrand™ 4.5-oz. Polypropylene Graduated Specimen Container). These removable containers, which can be filled with a preserving liquid such as propylene glycol or used without, and accompanying lids are used for labelling and storing collected specimens. Surrounding the apex of the chamber is a rounded collar that slopes gradually away from the opening (3 cm outward and 1 cm downward), with a steeper decline 0.5 cm from the outermost edge. The collar has raised ridges (0.1 mm high) spaced 1–2 mm apart to enable climbing by walking insects (Fig. 1A and D). Beneath the collar are four evenly spaced supports that link the collar to the chamber to provide rigid stability to the trap. At the apex of the collar are two 1.2-cm-diameter (outside diameter, OD) x 2-cm-high hollow wells, spaced 8.5 cm apart, which receive and secure the trap lid (Fig. 1A and D). The shape of the pitfall chamber is similar to typical hand-held or upright bulb planters, which can be used to quickly remove exact soil cores for tight trap insertion. Moreover, overlapping traps can be conveniently stacked for transport. When the base is inserted into the cored soil, foot pressure on the reinforced collar seals the base tightly to the

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ground (Fig. 1D). This process does not require the clearing of surface grass or excavation, as is typically required for other pitfall traps, and the raised collar helps reduce water entry into the base.

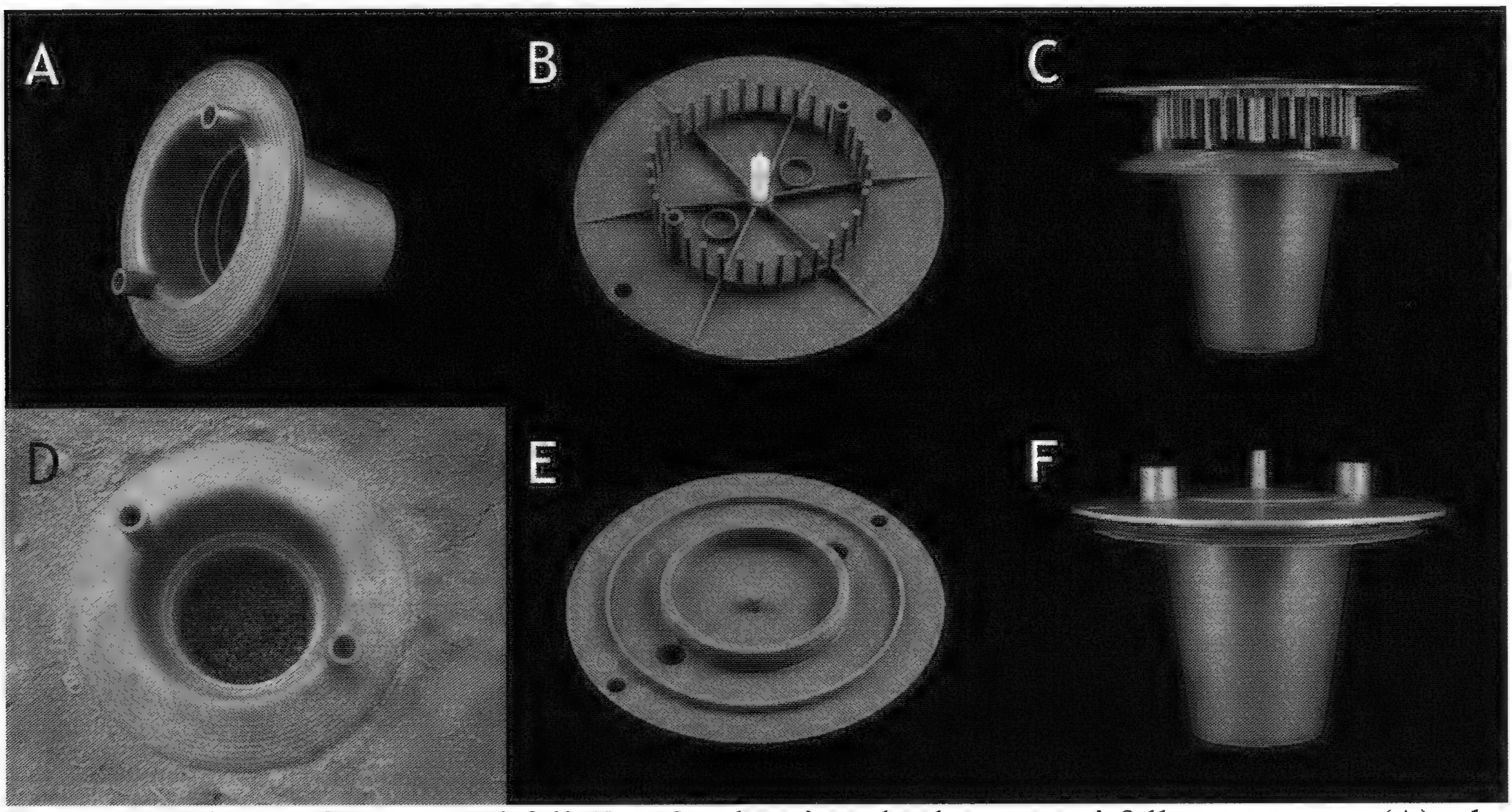

Figure 1. Views of Vernon Pitfall Trap®, showing the bottom, pitfall component (A), the underside of the cover with inserted lure and vertebrate exclusion fence (B), the assembled trap (C), the trap installed in soil with collected *A. sputator* (D), the optional cover for winterizing (E), and the assembled trap with winterizing cover (F). Photo credits: Warren Wong. Individual, high-resolution images are available as supplementary files on the journal website.

The second component is an easily detached cover, 16.5 cm in diameter, that tapers slightly downward as a shallow cone 0.5 cm from base to apex to shed rain (Fig. 1B). The cover contains a circular (1.2 cm diameter by 3 cm high, OD) downward-projecting well that is centred on the underside to hold 0.75-cm-diameter cylindrical *Agriotes* spp. pheromone baits (AO, AL and AS lures available from Csalomon, Budapest, Hungary), and two pegs (0.75-cm-diameter (OD) by 3 cm high) that are located 8.8 cm apart to fit into the corresponding wells on the base (Fig. 1A, B, and C). On the underside of the lid (Fig. 1B), six evenly spaced supports (0.5 cm high) that radiate from the projected pheromone lure well to the outside of the lid provide stability and prevent warping of the cover. To exclude insectivorous vertebrates (e.g., mice, voles, shrews, snakes) a circular fence of downward-projecting pins (3 mm diameter) is present on the underside of the lid. The pins are spaced 0.5 cm apart and range in length from 2.5 cm (30 pins) to 3 cm (4 pins). The longest pins just touch the base's collar section at four sites when base and lid are joined, lending stability to the assembled trap (Fig. 1C). The shorter pins leave a 0.5-cm-high passage above the collar to permit entry by click beetles or other walking insects.

The traps are manufactured in brown (used in Canada for AL), black (used for AO) and green (used for AS) to help avoid pheromone cross-contamination between species.

An optional trap component is a 16.5-cm-diameter winter lid (Fig. 1E) that replaces the main lid at the end of the trapping season. The winter lid is designed to snugly fit flush with the trap base (Fig. 1 F), so that the trap can remain *in situ* overwinter, protected from entry of debris, insects and water.

Should a need arise in the future, the cover's downward-projecting well for holding *Agriotes* pheromone lures could be replaced by a ring for hanging lures or a removable lure-holding basket, as in the Unitrap, for deploying lures for other target soil-surface

species. This would require the mold to be restructured. Alternatively, such lure-holders could be constructed as separate components that fit into the well.

The new trap offers a number of improvements to monitoring AO, AL and AS, relative to the former Vernon Beetle Trap. The VPT requires less time to assemble, install and inspect, and is more durable and transportable than the VBT. The VPT is similar to the VBT in catch of AO, AL and AS (van Herk, unpublished data). It has proven highly effective, with or without pheromone baits, in monitoring programs for AO and AL in BC and for AS in Prince Edward Island (PEI) (Table 1). The highest catch recorded to date for a single trap is 6,955 AS over a 5-day period in Orwell, PEI (27 May–1 June, 2015) (Fig. 1D). The trap has also been used, without pheromone baits, to successfully trap other elaterid species in other provinces of Canada, including *A. mancus* (Say), *Aeolus mellillus* (Say), *Hypnoidus abbreviatus* (Say), *H. bicolor* (Eschscholtz), *Limonius californicus* (Mannerheim), *Melanotus communis* (Gyllenhal), and *Selatosomus destructor* (Brown) (van Herk, unpublished data). It has also been used successfully to trap other walking insects, including carabids and weevils (e.g., *Sitona lineatus* L.; St. Onge *et al.* 2018).

Table 1

Catch of three *Agriotes* species in baited *versus* unbaited Vernon Pitfall Traps® (VPT) in field surveys in BC (AO and AL) and PEI (AS). N = number of traps.

Year	*Agriotes* Species[1]	Trapping period	Baited VPT		Unbaited VPT	
			N	Mean (SD)	N	Mean (SD)
2015	AO	26 Mar–16 July	22	977.7 (451.5)	33	10.2 (12.2)
2016	AL	21 Mar–11 July	22	171.0 (92.7)	33	0.8 (1.0)
2015	AS	20 May–13 Aug	44	7,797.1 (2,783.9)	38	71.6 (53.0)

[1]AO = *A. obscurus*; AL = *A. lineatus*; AS = *A. sputator*

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SCIENTIFIC NOTE

Identifying larval stages of *Orgyia antiqua* (Lepidoptera: Erebidae) from British Columbia, Canada

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The rusty tussock moth, *Orgyia antiqua* (Linnaeus, 1758) (Lepidoptera: Erebidae) is being used as an ecological surrogate to measure the impact of native natural enemies on the establishment of European gypsy moth, *Lymantria dispar* (Linnaeus, 1758), in British Columbia, Canada. To measure stage-specific mortality rates, one must be able to identify accurately different life stages of the species under study, ideally with characteristics that can be used in the field. The existing literature describing the number, size, and colouration of larval instars for *O. antiqua* is highly inconsistent (Table 1). The number of reported larval instars varies from 5–6 in males and 5–7 in females. Only two papers report the width of larval head capsules, with substantial disagreement between them (Dyer 1893; Payne 1917). Later instars of *O. antiqua* are characterised by dense tufts of setae on the dorsal surface of segments 4–7. These have been variously described as white, yellow, rusty brown, dark grey or black, and have been proposed by some authors to be aposematic warnings (Sandre *et al.* 2007a) that vary among instars. Through careful rearing of individual larvae and consistent measurements of head capsule width, we sought to clarify the number of larval instars and identify unique morphological characters that would facilitate the determination of instar in the field.

Table 1

Published descriptions of larval *O. antiqua* with respect to the colour patterns of the four dorsal tufts and the corresponding head capsule widths. Listed colours should be read as tuft colour from anterior to posterior starting on the first abdominal segment. B=Black, Y=Yellow, W=White, G=Grey, Br=Brown, ?=not reported.

	Dyer 1893	Gentner 1915	Hardy 1945	Payne 1917	Sandre 2007a	Sandre 2007b
Instar			Colour Pattern of Dorsal Tufts			
3	B-B-Y-Y	G-G-W-W	B-B-W-W	G-G-W-W	?-?-?-?	?-?-?-?
4	B-B-Y-Y	?-?-Y-Y	Y-Y-Y-Y	G-G-Y-Y	B-B-Y-Y (Pied)	B-B-Y-Y *
5	B-B-Y-Y	W-W-W-W	?-?-?-?	W-W-W-W	Y-Y-Y-Y (Bright)	Y-Y-Y-Y *
6	W-W-W-W	-	-	W-W-W-W	Br-Br-Br-Br (Dull)	Br-Br-Br-Br *
7	W-W-W-W	-	-	-	-	
			Head Capsule Width (mm)			
1	0.55			0.518 - 0.537		
2	0.75			0.812 - 0.875		
3	1.1			1.16 - 1.35		
4	1.55			1.80 - 2.02		
5	2.1			2.24 - 2.64		
6	-			3.0 - 3.5		
7	-			-		

*Sandre 2007b reports that this is the typical pattern but that "nearly all other combinations were also present."

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In May 2017, 33 *O. antiqua* egg masses were collected from a small population in Burnaby, BC, Canada (49.258821 N, 123.009661 W), that was feeding on an isolated Colorado spruce (*Picea pungens* Engelm) planted as a landscaping tree. Larvae were reared for one generation on *Alnus rubra*. In May 2018, 40 newly eclosed larvae from the second generation were reared individually in 50-mm plastic Petri dishes (20°C, 18L:6D) and fed fresh foliage of locally collected Himalayan blackberry (*Rubus armeniacus*) every 1–3 days. Head capsule widths for each larval instar were measured, using a Leica M5S dissecting microscope with an ocular micrometer with a precision of 0.012 mm, on live larvae that had been chilled for approximately 10 minutes at 5°C. Shed head capsules were retained for each individual larva in order to confirm the number of moults.

In a separate trial, a small sample of 10 newly eclosed *O. antiqua* larvae were reared on coastal Douglas-fir (*Pseudotsuga menziesii* var. *menziesii*) foliage to determine if they could complete development on this host. The head capsule widths for these larvae were measured for 4th and successive instars only. Representative photographs were taken of individual larvae from each instar using a Nikon D7000 digital camera equipped with a Nikon Speedlight SB-700 flash unit. After pupation and subsequent emergence, the gender of adults was recorded.

Of the 40 larvae reared on blackberry, eight died of unknown causes before the 3rd instar and were excluded from the analysis. Males (n = 17) invariably had five instars, whereas females (n = 15) typically had six instars, with the exception of one female that pupated after the 5th instar. For the first four instars, the head capsule widths of the larvae grew exponentially, closely following Dyar's rule (Fig. 1). For male and female larvae, 5th instar head capsules were smaller than expected, based on the progression of the first four instars. Similarly, head capsules of 6th instar females were also smaller than expected. There was no overlap in the head capsule widths of successive instars for either sex through 1st to 6th instar (Fig. 2), and our measurements closely matched those of Payne (1917). Larvae reared on Douglas-fir foliage had very similar head capsule widths to those reared on blackberry (Fig. 2). Consistent with the blackberry-reared larvae, the male larvae reared on Douglas-fir (n=5) had five instars, and the female larvae (n=5) predominantly had six instars with the exception of one female, which pupated following the 5th instar.

The morphological appearance of the first three instars closely matched the descriptions previously published in the literature (Table 1, Fig 3). First instar larvae are characterised by the absence of orange tubercles on the 6th and 7th abdominal segments. These tubercles are present in the 2nd instar larvae, but this stage lacks lateral pencils on the 1st thoracic segment. The 3rd instar is characterised by distinct lateral pencils on the 1st thoracic segment, as well as by the appearance of dorsal tufts on the 1st to 4th abdominal segments. We found that the dorsal tufts of 4th instar larvae always had a "pied" (Table 1) or two-toned colouration that varied considerably between individuals (e.g., larvae 6 and 7 in Fig. 3). Fifth instar males had four monochromatic tufts that ranged in colour from white to yellow to a rusty brown. In females that had six instars, the 5th instar was pied (e.g., Fig. 3, larvae 6 and 28). Sixth instar females looked the same as 5th instar males, with four monochromatic tufts ranging from white to yellow and rusty brown.

In conclusion, it is difficult to unambiguously discriminate between 4th, 5th, and 6th instars in the field based solely on the colouration of the dorsal tufts. An individual with tufts of different colours could be a 4th instar of either sex or a 5th instar female that will eventually moult into a 6th instar. An individual with tufts of a single colour could be a 5th instar of either sex or a 6th instar female. Head capsule width, however, could be used to discriminate unambiguously between each of the instars. In our sample, there was no overlap in the size distributions for each instar, even when we reared the larvae on Douglas-fir, which we considered a sub-optimal host based on previously observed slower growth rates.

It is interesting to note that when females had an 'extra' instar, it was not a typical supernumerary instar as has been reported in other lepidopteran larvae (Leonard 1970; Retnakaran, 1973; Hatakoshi *et al.* 1988) but rather a repetition of the 4th instar; the additional instar was morphologically similar to the 4th instar, only larger. Only one of the 15 females reared on blackberry did not have a 6th instar and that individual had the largest head capsule of all the female larvae from the 3rd to 5th instars. This suggests to us that the physiological trigger for an extra instar is related to size and that this is triggered at some point during or before the 4th instar.

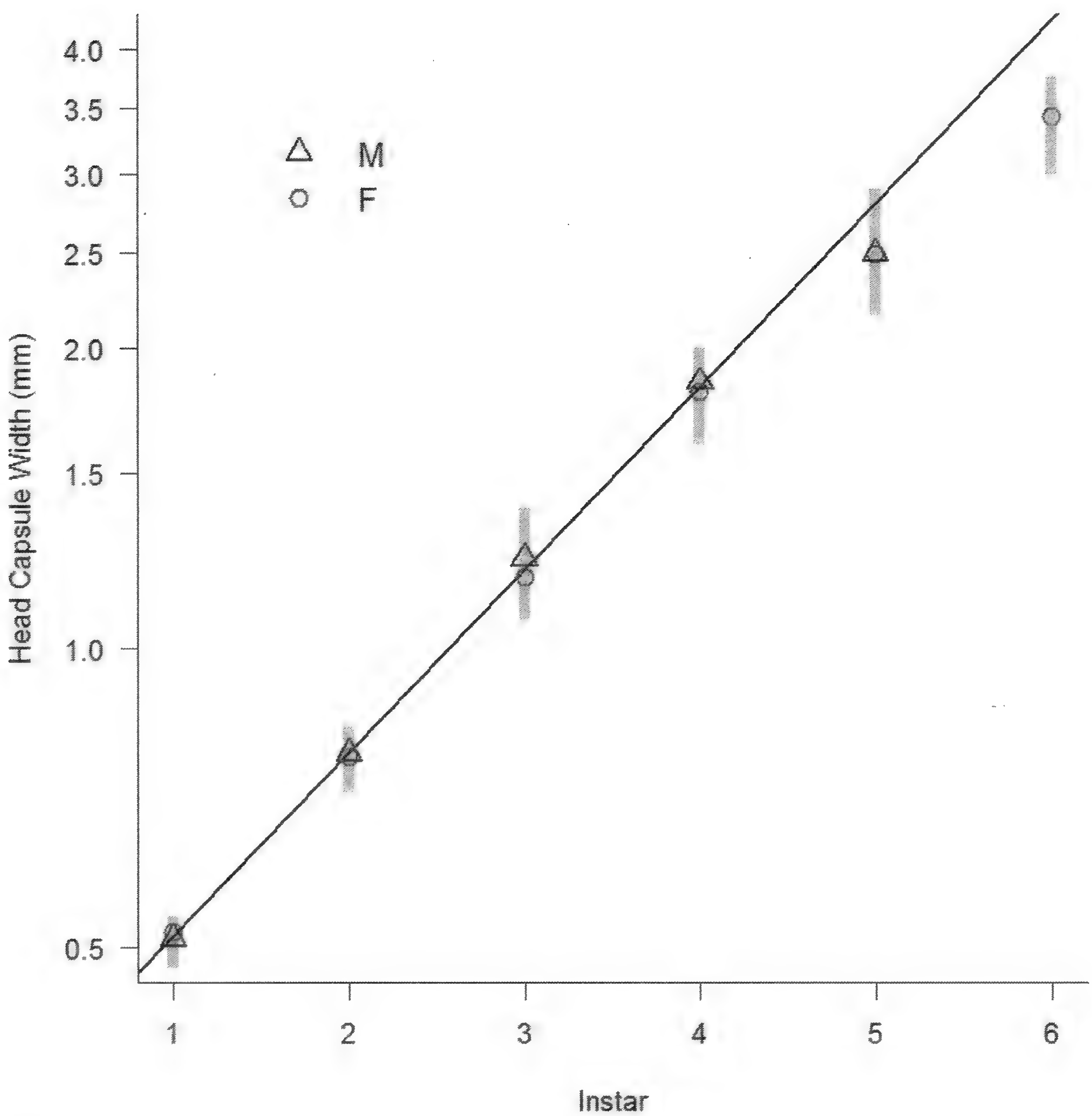

Figure 1. Average head capsule widths for female (circles) and male (triangles) *Orgyia antiqua* larvae according to instar number. The linear regression line was fitted to the first four instars only as the head capsule widths for the final two instars deviated significantly from a linear relationship $\log_{10}(HCW) = 0.185 \times Instar - 0.475$, ($R^2 = 0.999$, $F_{1,6}=7620$, $P<0.001$). Vertical grey bars represent the range of measurements for each instar.

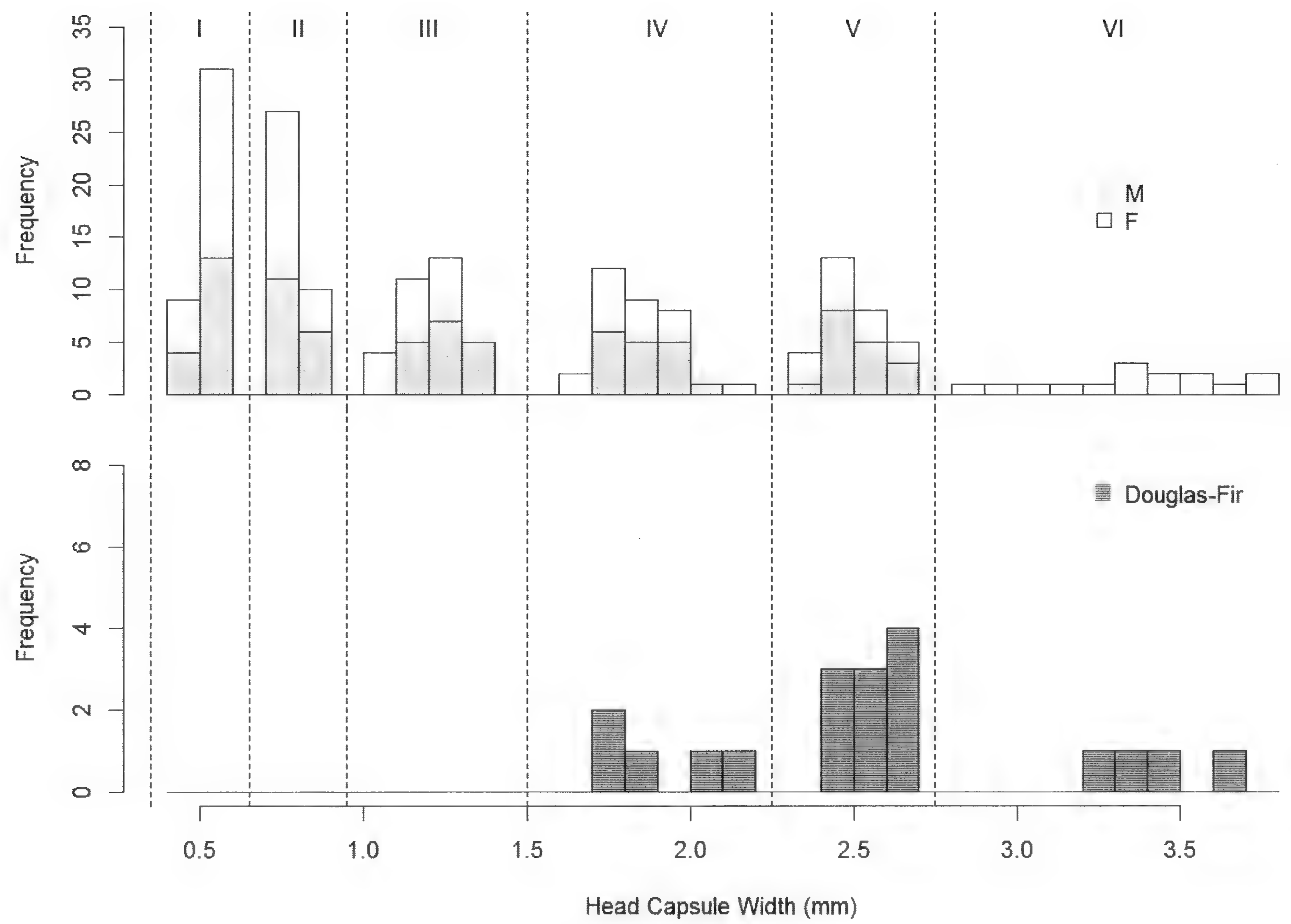

Figure 2. Distribution of head capsule sizes according to instar and sex when reared on Himalayan blackberry (top panel) and Douglas-fir (bottom panel). Instars were assigned based on the number of observed moults. Vertical dashed lines indicate proposed cut-off points to discriminate field collected larval instars.

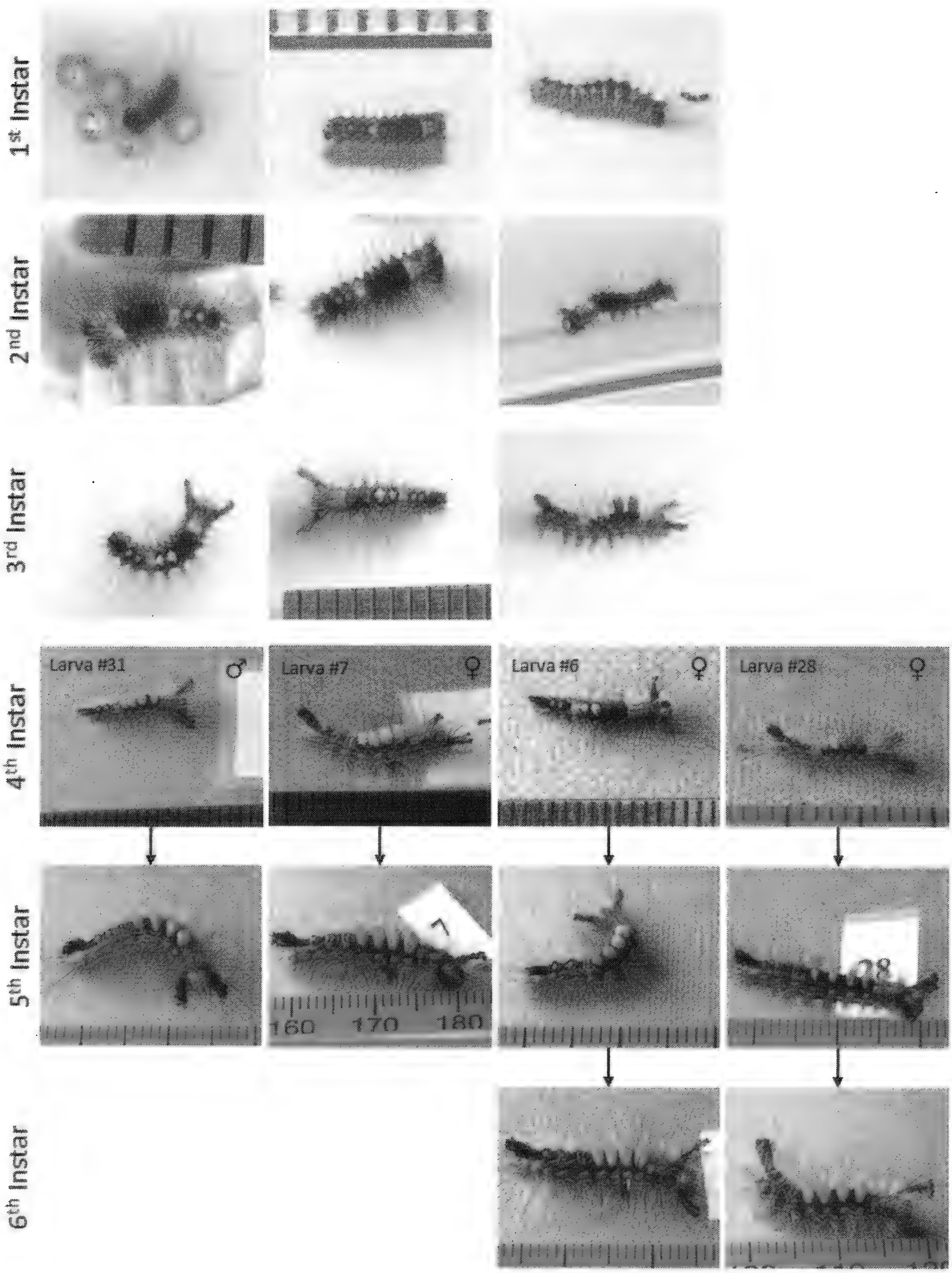

Figure 3. Representative images of *Orgyia antiqua* larvae reared on Himalayan blackberry leaves. Larvae in instars 1–3 (first three rows) exhibited little variation in colouration. The dorsal tufts were always uniformly coloured in the final instar, which was 5 in males and either 5 or 6 in females. Arrows between images indicate successive images of the same individual.

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NATURAL HISTORY AND OBSERVATIONS

New records of Hymenoptera from British Columbia and Yukon

C.G. RATZLAFF[1]

ABSTRACT— Thirty species of Hymenoptera are recorded for the first time from British Columbia and Yukon, including nine with records representing the first for Canada, with specimens from the families Bethylidae, Braconidae, Chrysididae, Crabronidae, Diapriidae, Figitidae, and Thynnidae. A description of the male of *Diodontus spiniferus* (Mickel) [Crabronidae], a correction to the distribution of *Dryudella elegans* (Cresson) [Crabronidae], and a correction to the locale for the holotype of *Aspicera mirieiae* Ros-Farré & Pujade-Villar [Figitidae] are also provided.

Key words: Hymenoptera, wasps, new, Canada, British Columbia, Yukon

INTRODUCTION

The diverse habitats of British Columbia and Yukon provide homes for a large number of insect species, including many that, in Canada, are found only in this area. Among the Hymenoptera, this is especially true, and new species are being recorded every year (Heron and Sheffield 2015; Ratzlaff 2015; Ratzlaff *et al.* 2016). Many groups of bees and wasps have been fairly well studied in British Columbia, while the last significant study of Yukon wasp fauna was Finnamore's chapter on aculeate wasps in the 1997 publication, *Insects of the Yukon*. Large swathes of remote wilderness cover much of the province and territory and, undoubtedly, many more known and unknown species have yet to be discovered. Even just recently, a new bumblebee species, *Bombus kluanensis* Williams & Cannings, was discovered in Yukon (Williams *et al.* 2016). Recent field collecting trips, along with study of existing museum specimens at the Spencer Entomological Collection, have resulted in 30 species of wasps being newly identified from British Columbia and Yukon. These records are presented here.

Collection abbreviations used are as follows: CGR – Author's personal collection; CNCI – Canadian National Collection of Insects, Arachnids, and Nematodes, Ottawa, ON; RBCM – Royal British Columbia Museum, Victoria, BC; SEM – Spencer Entomological Collection, Beaty Biodiversity Museum, University of British Columbia, Vancouver, BC. All specimens examined are located at the SEM with exception of two in the CGR and one in the RBCM. Unless otherwise indicated, all scale bars shown are equivalent to 1 mm.

FAMILY BETHYLIDAE

Anisepyris occidentalis (Ashmead)

First species records for Canada. Previously recorded from the western USA and Mexico (Gordh and Móczár 1990).

BRITISH COLUMBIA: 1♀, Galiano I., north end, 20.vii.1986 (G. G. E. Scudder) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 14.vi.–3.viii.1987, pan trap, *Purshia/ Aristida* steppe (S. G. Cannings) [SEM]; 1♂, Penticton, West Bench, 11.viii.1988, rose thicket/grassland boundary (S. G. Cannings) [SEM]; 2♂, Kalamalka Lake Prov. Pk., 21.viii.1987 (S. G. Cannings) [SEM] (Fig. 1); 1♀, Osoyoos, East Bench, 28.v.2000, biting person (J. Scudder) [SEM]; 1♂, Tsawwassen, Boundary Bay Regional Pk., 49.0176°N, 123.0422°W, 10.viii.2015 (C. G. Ratzlaff) [SEM]

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Figure 1. Male *Anisepyris occidentalis,* from Kalamalka Lake Provincial Park, BC.

***Epyris clarimontis* Kieffer**

First species records for Canada. Recorded as being widespread in the USA and Mexico (Gordh and Móczár 1990).

BRITISH COLUMBIA: 2♀, Osoyoos, Haynes Ecological Reserve, 20.v.–14.vi. 1987, pan trap, *Purshia/Aristida* steppe (S. G. Cannings) [SEM]; 3♀, Osoyoos, Haynes Ecological Reserve, 14.vi.–3.viii.1987, pan trap, *Purshia/Aristida* steppe (S. G. Cannings) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 13.vii.–17.viii.1988, pitfall trap, *Purshia/Aristida* steppe (S. G. Cannings) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 23.vii.–26.viii.1989, pitfall trap, rose thicket (S. G. Cannings) [SEM]; 1♂, Osoyoos, Haynes Ecological Reserve, 26.viii.–23.ix.1989, pitfall trap, *Rosa* clump at edge of wetland (S. G. Cannings) [SEM]

FAMILY BRACONIDAE

***Ascogaster borealis* Shaw**

First species record for Yukon. Previously recorded from BC, SK, ON, QC, NS, WA, ID, MT, WI, and ME (Shaw 1983).

YUKON: 1♂, Million Dollar Falls, 60.1082°N, 136.9466°W, 26.vi.2017 (SEM Team) [SEM]

***Meteorus vulgaris* (Cresson)**

First species record for Yukon. Previously recorded from all of southern Canada and much of the USA (Muesebeck 1923).

YUKON: 1♀, Carcross, Montana Mt., 60.1341°N, 134.7195°W, 28.vi.2017, 1075 m (SEM Team) [SEM]

FAMILY CHRYSIDIDAE

***Chrysurissa densa* (Cresson)**

First species records for Canada. Previously recorded from the western half of the USA (Kimsey 2005).

BRITISH COLUMBIA: 1♂, SOCAP Site #28, 15.v.1990 (H. Knight) [SEM]; 1♂, Osoyoos, Haynes Ecological Reserve, 1.vi.2000 (G. G. E. Scudder) [SEM]

***Pseudospinolia neglecta* Shuckard**

First species record for British Columbia. Previously recorded from AB, WA, CO, MT, MN, NE, and NY. It is also found in the Palearctic region (Bohart and Kimsey 1982).

BRITISH COLUMBIA: 1♀, Attachie, Don Phillips Way (Hwy. 29), 10V 599090 6233848 (56.23917°N, 121.40123°W), 22.vi.2015, 631 m (C & D Copley, J. Heron, H. Gartner & K. Ovaska) [RBCM] (Fig. 2)

Figure 2. Female *Pseudospinolia neglecta,* from Attachie, BC.

FAMILY CRABRONIDAE

***Crabro nigrostriatus* Bohart**

First species record for Yukon. Previously recorded from BC, OR, NV, and CA (Bohart 1976).

YUKON: 1♂, Kookatsoon L., 60.5587°N, 134.8758°W, 29.vi.2017 (SEM Team) [SEM] (Fig. 3)

***Diodontus argentinae* Rohwer**

First species records for Yukon. Previously recorded from BC, WA, OR, WY, CA, CO, UT, DC, and Mexico (Eighme 1989).

YUKON: 1♂, Kluane Nat. Pk., Sheep Mt., 5.vii.1979 (S. G. Cannings) [SEM]; 1♂, Pelly Crossing, 2.vii.1985 (E. Bijdemast) [SEM]; 1♂, Dawson, 13.vii.1985, steep Artemesia slope (S. G. Cannings) [SEM]

***Diodontus bidentatus* Rohwer**

First species records for Yukon. Previously recorded from BC, AB, QC, NB, AK, ID, MT, CO, NE, NY, MI, ND, and PA (Krombein 1979; Eighme 1989; Finnamore 1994; Buck 2004; Ratzlaff 2015).

YUKON: 1♂, Duke River, Burwash Landing, 9.vii.1979 (S. G. Cannings) [SEM]; 1♂, Kluane L., Emerald I., 61.0209°N, 138.4893°W, 24.vi.2017 (SEM Team) [SEM]

Figure 3. Male *Crabro nigrostriatus,* from Kookatsoon Lake, YT.

***Diodontus leguminiferus* Cockerell**

First species records for Yukon. Previously recorded from BC, AB, ID, CA, MT, CO, UT, AZ, NM, MO, and IA (Eighme 1989; Ratzlaff 2015).

YUKON: 1♂, Carcross, sand dunes, 20.vii.1987 (S. G. Cannings) [SEM]; 1♂, Carcross Desert, 60.1876°N, 134.6899°W, 30.vi.2016 (C. G. & N. A. Ratzlaff) [SEM]; 2♂, Carcross Desert, 60.1876°N, 134.6899°W, 28.vi.2017 (SEM Team) [SEM]

***Diodontus occidentalis* Fox**

First species records for Yukon. Previously recorded from BC, AB, AK, ID, CA, NV, UT, WY, CO, AZ, MI, NY, and ND (Eighme 1989; Finnamore 1994; Ratzlaff 2015).

YUKON: 1♀, Silver City, 23.vii.1979 (G. G. E. Scudder) [SEM]; 1♀, Pelly Crossing, 26.vii.1980 (R. J. Cannings) [SEM]; 1♂, Tenas Creek, 5 km East on North Canol Rd., 62°02'N 132°14'W, 11.vi.1981 (C. S. Guppy) [SEM]; 1♀, Haines Junction, Pine Cr., 25.vi.1981 (C. S. Guppy) [SEM]; 1♀, Porcupine R., Blue Bluffs, 67°38'N 138°38'W, 11.vii.1981 (C. S. Guppy) [SEM]; 1♂1♀, Old Crow, 6 km E, 67°34'N 139°41'W, 13.vii.1981 (C. S. Guppy) [SEM]; 1♀, Whitehorse, Wolf Cr., 17.vii.1981 (C. S. Guppy) [SEM]; 1♀, Slims R. delta, 21.vi.1982 (R. D. Wilkie & S. G. Cannings) [SEM]; 1♀, Kluane, Sheep Mt., 24.vi.1982 (S. G. Cannings, R. D. Wilkie, L. Vasington & R. A. Moore) [SEM]; 1♀, Carmacks, 30 km E, 62°02'N 135°51'W, 10.vii.1982 (S. G. Cannings, L. Vasington & R. A. Moore) [SEM]; 1♀, Old Crow, 30.vi.1983, top of open S-facing bluff, malaise trap (R. A. Cannings) [SEM]; 1♀, Old Crow, 2.vii.1983, top of open S-facing bluff, malaise trap (R. A. Cannings) [SEM]; 1♀, Old Crow, 4.vii.1983, top of open S-facing bluff, malaise trap (S. G. Cannings) [SEM]; 1♀, Little Salmon L., 35 km E, 28.vi.1985 (E. Krebs & J. J. Robinson) [SEM]; 2♀, Tatchun L., 29.vi.1985 (E. Krebs & J. J. Robinson) [SEM]; 2♂, Pelly Crossing, 2.vii.1985 (S. G. Cannings) [SEM]; 1♀, Dawson, Midnight Dome, 12.vii.1985 (E. Bijdemast) [SEM]; 1♂, Carcross, Montana Mt., 60.1341°N, 134.7195°W, 28.vi.2017, 1075m (SEM Team) [SEM]

***Diodontus spiniferus* (Mickel)**

First species records for British Columbia and Yukon. Previously recorded from AB, ON, QC, CA, MT, CO, IA, NE, MD, MO, and MN (Eighme 1989; Buck 2004). The male of the species has never been described, but a few key characters were provided by Buck (2004) that are useful when comparing it to eastern specimens. Several other similar species exist in western Canada, and the necessary characters for identification are described here.

Male. (Fig. 4) Black. Mandible (except base and teeth), palps, apex of fore-femur, fore- and mid-tibiae dorsally, and anterior half of tegula yellow. Hind-tibia brownish-yellow dorsally, fading apically or nearly all brown in darker specimens. Fore and mid-tarsi yellow, hind-tarsi brown, the last two segments of all tarsi darkened. Antenna with small placoids on flagellomeres V–X, ranging from reddish-brown to brown. Frons with numerous larger punctures, often with much reticulation, giving it a rough appearance. Humeral angle prominent with close to a 90° angle. Propodeum reticulate. Wing veins and stigma brown. Abdominal terga sparsely punctate, lightly reticulate.

Figure 4. Male *Diodontus spiniferus*, from Kluane National Park, YT.

Using Eighme's (1989) key, males of *D. spiniferus* end up at couplet 16 with *retiolus* and *leguminiferus* but lack the strong reticulation on the abdomen found in *retiolus*. They differ from *leguminiferus* in having numerous large punctures and stronger sculpture on the frons (Fig. 5a), a prominent, roughly 90° humeral angle (Fig. 5b), and reddish-brown placoids on the antenna (Fig. 5c). Two other similar species are *boharti* and *crassicornus,* but *spiniferus* differs from the former by having a dark pronotal lobe and from the latter by having much less-inflated antenna and smaller placoids.

BRITISH COLUMBIA: 1♂, Pink Mt., 24 km S, 24.vi.1985 (S.G. Cannings) [SEM]

YUKON: 3♂2♀, Kluane Nat. Pk., Sheep Mt., 8.vi.1979 (S. G. Cannings) [SEM] (Fig. 4; Fig. 5c); 1♂1♀, Carmacks, Mt. Nanson Rd., 62.0587°N, 136.3781°W, 26.vi.2016 (C. G. & N. A. Ratzlaff) [SEM] (Fig. 5a, b); 1♀, Ibex Valley Salt Flats, 60.8616°N, 135.7126°W, 23.vi.2017 (SEM Team) [SEM]

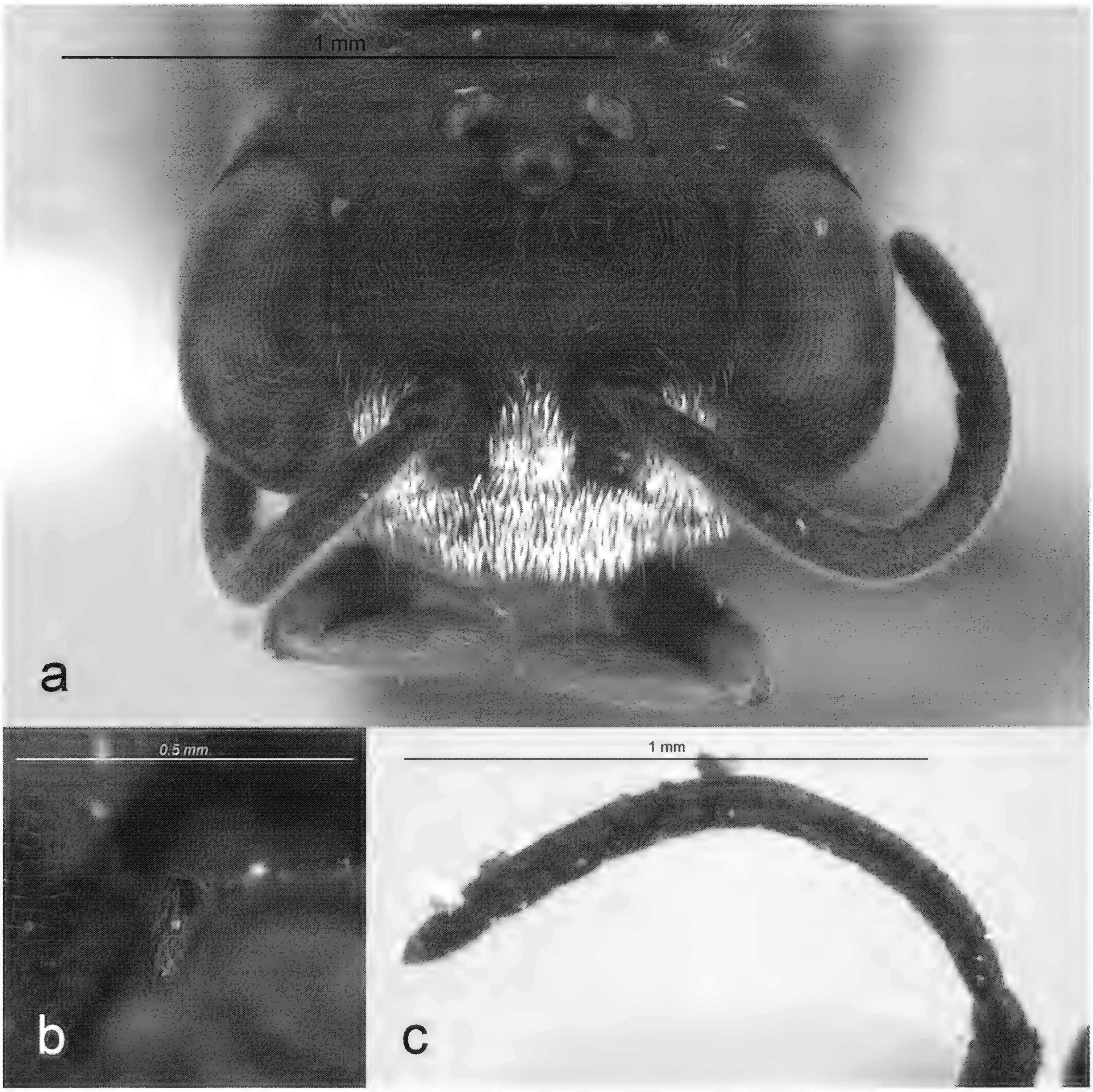

Figure 5. The face (a) and posterolateral view of the humeral angle on the pronotum (b) of a male *Diodontus spiniferus* from Carmacks, YT. The antennal placoids (c) of a male *D. spiniferus* from Kluane National Park, YT.

***Diodontus vallicolae* (Rohwer)**

First species record for Yukon. Previously recorded from BC, AB, AK, ID, WY, CA, CO, NV and UT (Eighme 1989).

YUKON: 1♂, Carcross, sand dunes, 20.vii.1987 (S. G. Cannings) [SEM]

***Dryudella elegans* (Cresson)**

In Cresson's (1881) original species description for *D. elegans* (as *Astata elegans*), the holotype location is given as "Washington Territory" and the paratype locations as "Vancouver's Island", Nevada, and Colorado. These locations appear again in Fox's (1893) synopsis of the North American Larridae and then disappear from all subsequent publication on the species, with the exception of Nevada. Parker (1969) records *D. elegans* from ID, WY, UT, NV, CA, and AZ, stating the holotype location only as "W. T." It appears that *D. elegans* should also be listed from BC (Vancouver Island), WA, and CO even thouth it currently is not. Why these localities were not included in the subsequent published species distributions is unknown, but additional British Columbian records are presented here, confirming the original northern range.

BRITISH COLUMBIA: 1♂, Osoyoos, Haynes Ecological Reserve, 9.vii.–9.viii. 1996, BGxh1, pitfall trap (G. G. E. Scudder) [SEM]; 1♂, Oliver, McKinney Rd., 49.19869°N, 119.49967°W, 26.vii.2017 (C. G. Ratzlaff) [CGR]

***Philanthus pulcher* Dalla Torre**

First species record for Yukon. Finnamore (1997) expected this species to occur in the territory, and it has been previously recorded from the western half of Canada and the USA, including NT (Bohart and Grissell 1975).

YUKON: 1♂, Pelly Crossing, 2.vii.1985 (S. G. Cannings) [SEM]

***Solierella albipes* (Ashmead)**

First species record for Canada. Previously recorded from ID, CO, and CA (Krombein 1979).

BRITISH COLUMBIA: 1♀, Osoyoos, Strawberry Creek Rd., 49.0364°N, 119.5002°W, 9.viii.2016 (C. G. Ratzlaff) [SEM] (Fig. 6)

Figure 6. Female *Solierella albipes,* from Osoyoos, BC.

***Solierella sayi* (Rohwer)**

First species records for Canada. Previously recorded from CO and CA (Krombein 1979).

BRITISH COLUMBIA: 2♂, Whipsaw Creek Forest Service Rd., 49.3536°N, 120.6097°W, 7–10.viii.2016, 986m, blue pan (C. G. Ratzlaff) [CGR, SEM]

FAMILY DIAPRIIDAE

***Ismarus halidayi* Förster**

First species record for British Columbia. Previously recorded in the Nearctic region from AB, NB, NF, CA, and MO, and in the Palearctic region from England and Finland (Masner 1976).

BRITISH COLUMBIA: 1♀, Sidney I., Dragonfly Pond, 49.6033°N, 123.3046°W, 14.viii.2016 (SEM Team) [SEM]

FAMILY FIGITIDAE

***Alloxysta halterata* (Thomson)**

First species records for Canada. Previously recorded in the Nearctic region from CO, and in the Palearctic region from England, Finland, Germany, Scotland, and Sweden (Ferrer-Suay *et al.* 2014; Ferrer-Suay 2017).

YUKON: 1♂, White Mts., "Erebia Cr.", 67°58'N 136°29'W, 2.vii. – 9.vii.1987, 2500', sandstone slope, pan trap (S. G. Cannings) [SEM] (Fig. 7); 1♀, Emerald L., 60.2639°N, 134.7520°W, 29.vi.2017 (SEM Team) [SEM].

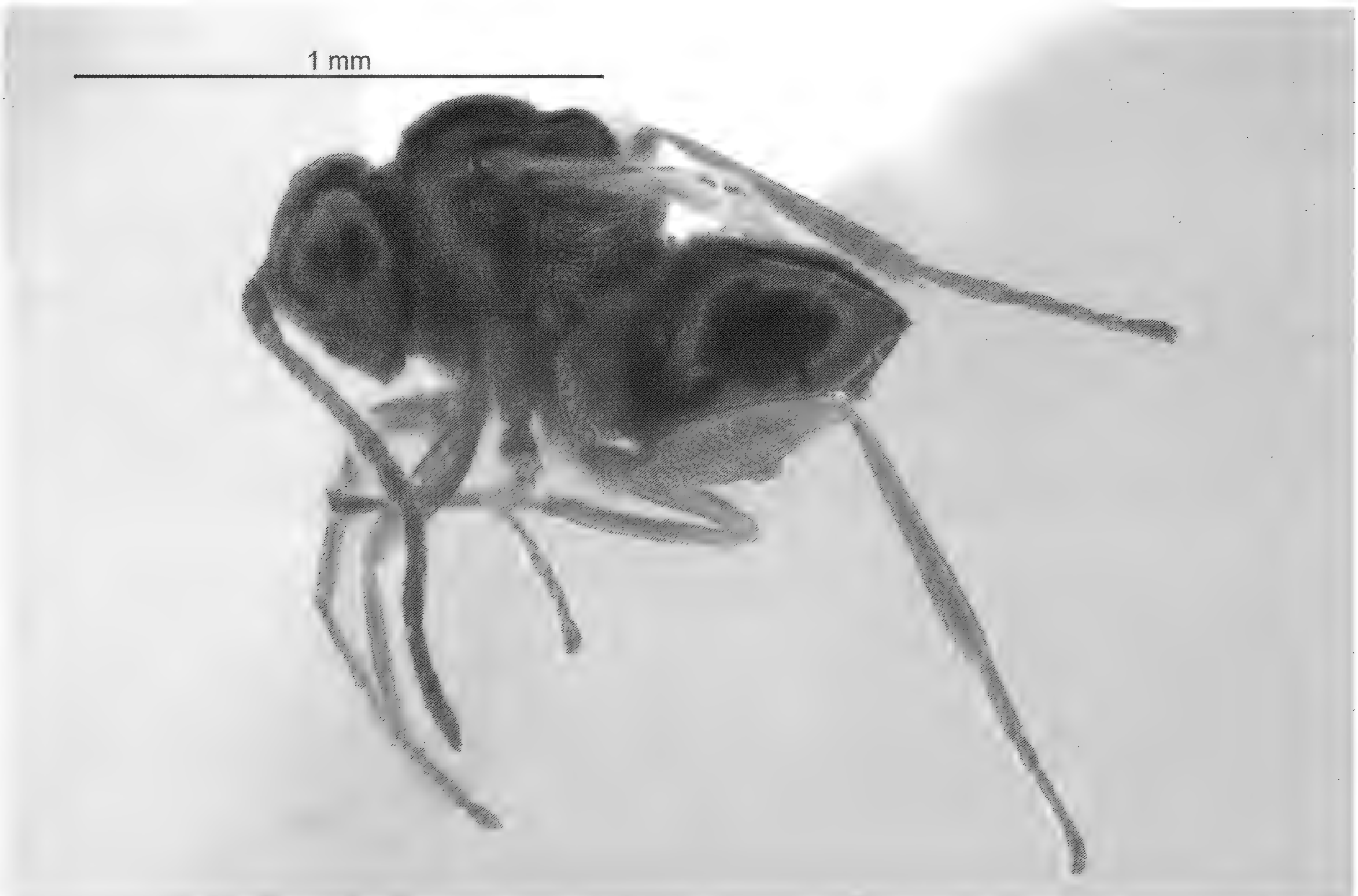

Figure 7. Male *Alloxysta halterata,* from the White Mountains, YT.

***Alloxysta obscurata* (Hartig)**

First species record for Yukon. Previously recorded in the Nearctic region from BC and AK, and in the Palearctic region from Andorra, France, Germany, Hungary, Iceland, Poland, Romania, and Scotland (Ferrer-Suay 2017).

YUKON: 1♀, Cottonwood Cr., 60°55'N 132°58'W, 2.viii.1981 (C. S. Guppy) [SEM]

***Alloxysta pallidicornis* (Curtis)**

First species record for British Columbia. Previously recorded in the Nearctic region from AB, QC, AK, and CO, and in the Palearctic region from Austria, England, Finland, France, Germany, Norway, Spain, and Sweden (Ferrer-Suay 2017).

BRITISH COLUMBIA: 1♀, Saturna I., Gulf Islands National Pk. & Reserve, 48.8084°N, 123.1856°W, 17.vii.2015 (C. G. Ratzlaff) [SEM]

***Alloxysta postica* (Hartig)**

First species records for Canada. Previously recorded in the Nearctic region from AZ and in the Palearctic region from Bulgaria, Czech Republic, and Germany (Ferrer-Suay *et al.* 2014; Ferrer-Suay 2017).

YUKON: 1♀, Emerald L., 60.2639°N, 134.7520°W, 29.vi.2017 (SEM Team) [SEM]; 1♀, Kookatsoon L., 60.5587°N, 134.8758°W, 29.vi.2017 (SEM Team) [SEM]

***Aspicera mirieiae* Ros-Farré & Pujade-Villar**

The location data for the HOLOTYPE is wrongly stated in the original species description (Ros-Farré and Pujade-Villar 2013). In the publication, the record is listed as "HOLOTYPE male (CNCI) 27/VII/1959, Summit L. Mi392, 420' Alaska USA, Hwy B.C., E. E. MacDougall leg.", and is listed as an Alaskan and American locale. The actual label and location should be read as "BC, Alaska Hwy., mi. 392, Summit L.", making this a British Columbian locale and Canadian record. The approximate coordinates of mile 392 on the Alaska Highway, which originates in Dawson Creek, BC, would be 58.8499°N, 125.0617°W. This correction was determined by examining specimen labels with the same location and collector, with nearby dates, present in the SEM collection.

BRITISH COLUMBIA: 1♂, BC, Alaska Hwy., mi. 392, Summit L., 27.vii.1959, 4200' (E. E. MacDougall) [CNCI]

***Aspicera santamariai* Ros-Farré & Pujade-Villar**

First species records for British Columbia and Yukon. Previously recorded from AB (Ros-Farré and Pujade-Villar 2013).

BRITISH COLUMBIA: 1♀, Penticton, West Bench, 6.vi.1988 (S.G. Cannings) [SEM]

YUKON: 2♂ 2♀, Carcross, sand dunes, 20.vii.1987 (S. G. Cannings) [SEM] (Fig. 8); 1♀, Silver City, 61.0480°N, 138.3878°W, 24.vi.2017 (SEM Team) [SEM]

Figure 8. Male *Aspicera santamariai,* from Carcross, YT.

***Omalaspis cavroi* (Hedicke)**
First species record for Yukon. Previously recorded from BC, AB, ON, QC, NB, AK, MT, CA, AR, and ME (Ros-Farré & Pujade-Villar 2011b).
YUKON: 1♂, Carcross, sand dunes, 20.vii.1987 (S. G. Cannings) [SEM]

***Paraspicera brandaoi* Ros-Farré & Pujade-Villar**
First species records for Yukon. Previously recorded from BC, AB, and ID (Ros-Farré and Pujade-Villar 2011a).
YUKON: 1♂, Old Crow, 1 km E, 16.vii.1981 (C.S. Guppy) [SEM]; 1♂, Old Crow, 2.vii.1983, top of open S-facing bluff, malaise trap (S. G. Cannings) [SEM]

***Phaenoglyphis gutierrezi* Andrews**
First species record for Yukon. Previously recorded from BC, SK, and MT (Andrews 1978).
YUKON: 1♀, Cottonwood Cr., 60°55'N 132°58'W, 2.viii.1981 (C. S. Guppy) [SEM]

***Phaenoglyphis pilosus* Andrews**
First species record for Yukon. Previously recorded from BC, AB, ID, CA and CO (Andrews 1978).
YUKON: 1♀, Emerald L., 60.2639°N, 134.7520°W, 29.vi.2017 (SEM Team) [SEM]

***Phaenoglyphis ruficornis* (Förster)**
First species record for Yukon. Previously recorded in the Nearctic region from BC, SK, ON, QC, and CA, and in the Palearctic region from Germany and Israel (Ferrer-Suay 2017).
YUKON: 1♀, Tagish, 22.vii.1981 (S. G. Cannings) [SEM]

***Phaenoglyphis villosa* (Hartig)**
First species record for Yukon. A very widespread species that has been recorded from every continent except Antarctica (Ferrer-Suay 2017).
YUKON: 1♀, Kluane Nat. Pk., S end of Kluane L., 60.9930°N, 138.4674°W, 24.vi. 2017 (SEM Team) [SEM]

***Sarothrus nasoni* Ashmead**
First species record for Canada. Previously known only from IL (Burks 1979).
BRITISH COLUMBIA: 1♀, Pink Mt., 57.0487°N, 122.8687°W, 2.vii.2016, 1715m (C. G. & N. A. Ratzlaff) [SEM] (Fig. 9)

FAMILY THYNNIDAE
Lalapa lusa Pate
First species records for Canada. Goulet and Huber (1993) suspected that this species occurred in southern BC, and it has been previously recorded from WA, ID, OR, and CA (Johnson *et al.* 1995).

BRITISH COLUMBIA: 1♀, Osoyoos, Haynes Ecological Reserve, The Throne, 10.vii.–14.viii.1986, under sage brush, pitfall trap (S. G. Cannings) [SEM]; 1♀, Penticton, West Bench, 3.viii.1987 (S. G. Cannings) [SEM]; 1♀, Penticton, West Bench, 23.viii.1987 (S. G. Cannings) [SEM] (Fig. 10); 1♀, Osoyoos, Haynes Ecological Reserve, 13.vii.–17.viii.1988, Purshia/Aristida steppe, pitfall trap (S. G. Cannings) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 9.viii.1995, (G.G.E. Scudder) [SEM]; 1♀, Osoyoos, Haynes Ecological Reserve, 15.viii.–11.ix.2004, BGxh1, AN Recovery after 1993 fire, Pitfall trap ER2-4 (G. G. E. Scudder) [SEM]

Figure 9. Female *Sarothrus nasoni,* from Pink Mountain, BC.

Figure 10. Female *Lalapa lusa,* from Penticton, BC.

CONCLUSION

Bioblitzes have become an important part of the study of flora and fauna in British Columbia, the Yukon Territories, and the rest Canada. These events facilitate a concentrated effort to document the species present in areas where often not much has previously been done. This is particularly true in places with regulated research access, such as national parks and, as a result, the known range of many species has been expanded. Much of this new material, however, unfortunately ends up unidentified in different natural history collections, alongside many other unexamined specimens. Undoubtedly, study of these specimens will yield new information about many species in British Columbia and the Yukon.

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Thank you to Syd Cannings, Parks Canada, and the other organizers of the 2016 Carmacks Bioblitz and the 2017 Kluane National Park Bioblitz for the invitation and the opportunity to visit these unique Yukon habitats. Thank you to Athena George, Parks Canada, and the other organizers of the 2015 Saturna Island Bioblitz and the 2016 Sidney Island Bioblitz for the invitation and opportunity to visit the Gulf Islands National Park and Reserve.

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NATURAL HISTORY AND OBSERVATIONS

First Record of *Culex tarsalis* (Diptera: Culicidae) in the Yukon

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ABSTRACT— The first record of *Culex tarsalis* in the Yukon is reported from a larva collected in Kluane National Park in 2017. Details on the location and the specimen are provided, and background information on the biology of *Cx. tarsalis* and its role in arbovirus transmission are discussed.

Key words: *Culex tarsalis,* Western encephalitis mosquito, Culicidae, Yukon, mosquito distribution

The western encephalitis mosquito, *Culex tarsalis* Coquillett, is a medium-sized mosquito (wing length 4.0-4.4 mm) with bands of white scales on the tarsi and a broad ring of white scales on the proboscis at mid-length (Belton 1983). Adult females overwinter in sheltered areas such as caves, rodent burrows, and under rock piles (Wood *et al.* 1979), and larvae are found in a wide variety of habitats including ponds, marshes, ditches, and irrigation water (Belton 1983). Adults have been observed feeding from flowers of goldenrod (*Solidago spp.*) (Sandholm and Price 1962) and common tansy (*Tanacetum vulgare*), from which they carry pollen (Peach and Gries 2016). Females take blood from birds and mammals (Wood *et al.* 1979).

Cx. tarsalis is an important vector of several viruses in southern Canada, including West Nile virus (Roth *et al.* 2010, Kulkarni *et al.* 2015), western equine encephalitis (McLintock *et al.* 1970), and St. Louis encephalitis (Hammon and Reeves 1943), although these are not known from the Yukon (Artsob 1990). Snowshoe hare virus, in the California Encephalitis (CE) group, is endemic in the Yukon (McLean *et al.*1973) but is not reported to have been isolated from *Cx. tarsalis*. However, CE itself has been isolated from *Cx. tarsalis* in California (Hammon *et al.* 1952). Northway virus is also endemic to the Yukon (McLean and Lester 1983), but little is known about this virus or its vectors.

The known range of *Cx. tarsalis* extends throughout much of central and western North America (Darsie and Ward 2005), including southern British Columbia (Belton 1983) and southern Alberta (Wood *et al.*1979). It has also been found in Norman Wells, (65°N) in the Northwest Territories (Freeman 1952) and Belton and Belton (1990) believed the species was likely to occur in the Yukon as well, based on its inclusion in a list of Yukon mosquitoes by Nelson (1977). Nelson cites a personal communication from D. M. Wood to support this, but Wood *et al.* (1979) show no records of *Cx. tarsalis* in the Yukon.

A *Culex* sp. larva was collected in a shallow pond in Kluane National Park, Yukon, Canada just outside the Slims River Flats (60°59'23.6"N, 138°29'31.9"W) on 24 June, 2017 as part of the Kluane Park bioblitz (research permit number KLU-2017-25041). The larva was successfully reared to adulthood, and the female was identified as *Cx. tarsalis* (Fig. 1) using the key of Wood *et al.* (1979). This specimen represents the first confirmed record of this species in the Yukon. Of note is the incomplete white-scaled ring at midpoint of the proboscis of this specimen as it possesses dark scales dorsally, possibly due to phenotypic plasticity related to thermal melanism (Trullas *et al.* 2007) or poor larval habitat conditions (Talloen *et al.* 2004). The pond was adjacent to the Alaska Highway, approximately 10 metres in diameter, shallow, and contained clear water.

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Nearby vegetation included willow (*Salix spp.*), spruce (*Picaea sp.*), fireweed (*Chamaenerion sp.*), and patches of unidentified grass. Larvae of *Anopheles earlei*, *Aedes excrucians,* and *Culiseta alaskaensis* were also collected from the pond, and adults captured nearby included *Ae. campestris*, *Ae. cataphylla, Ae. communis, Ae. excrucians*, *Ae. fitchii*, and *Ae. implicatus*. The *Cx. tarsalis* specimen has been deposited for reference in the Beaty Biodiversity Museum at the University of British Columbia, Vancouver, British Columbia. Due to the short summer season and the likeliness that only small populations may be present it seems unlikely that *Cx. tarsalis* currently poses a major human health risk in the Yukon. However, if temperatures rise these limiting conditions may no longer apply (Chen *et al.* 2013).

Figure 1. Antero-dorsal (A) and lateral (B) views of the *Cx. tarsalis* specimen collected in the Yukon. Note incomplete band of white scales at mid-proboscis in (A), indicated by an arrow.

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NATURAL HISTORY AND OBSERVATIONS

An updated list of the mosquitoes of British Columbia with distribution notes

DANIEL A.H. PEACH[1]

Since "The Mosquitoes of British Columbia", originally published by Dr. Peter Belton 35 years ago, there have been only sporadic and incomplete updates on the mosquito fauna of British Columbia (BC). Darsie and Ward's (2005) "Identification and Geographical Distribution of the Mosquitoes of North America, North of Mexico" reported the presence and distribution of many species within BC but was continent-wide in scope and did not provide BC-specific information. It also disregarded the presence or distribution of several species.

Belton (1983) recognized 46 mosquito species as occurring within British Columbia, discounting previous records of *Culex restuans* but including *Aedes nevadensis* due to specimens he had collected from the BC Interior. Darsie and Ward (2005) recognized 45 species, discounting records of *Ae. nevadensis* from Belton (1983) as well as previous records of *Cx. restuans*. Since 2005, several additional species records have been made for BC, and a new record of *Cx. restuans* from southern Vancouver Island supports its inclusion as part of BC's mosquito fauna, bringing the total number of species known from the province to 50. In several instances, the distribution of various species within BC has also been extended, due to new collection records in previous unsurveyed or undersurveyed areas.

List of the mosquito species known from British Columbia

Aedes (*Ochlerotatus*) *aboriginis* Dyar
Aedes (*Ochlerotatus*) *aloponotum* Dyar (Updated distribution)
Aedes (*Ochlerotatus*) *campestris* Dyar & Knab
Aedes (*Ochlerotatus*) *canadensis* (Theobald)
Aedes (*Ochlerotatus*) *cataphylla* Dyar
Aedes (*Aedes*) *cinereus* Meigen (Updated distribution)
Aedes (*Ochlerotatus*) *communis* (De Geer)
Aedes (*Ochlerotatus*) *diantaeus* Howard, Dyar & Knab
Aedes (*Ochlerotatus*) *dorsalis* (Meigen)
Aedes (*Ochlerotatus*) *euedes* Howard, Dyar & Knab
Aedes (*Ochlerotatus*) *excrucians* (Walker)
Aedes (*Ochlerotatus*) *fitchii* (Felt & Young)
Aedes (*Ochlerotatus*) *flavescens* (Müeller)
Aedes (*Ochlerotatus*) *hendersoni* Cockerell
Aedes (*Ochlerotatus*) *hexodontus* Dyar
Aedes (*Ochlerotatus*) *impiger* (Walker)
Aedes (*Ochlerotatus*) *implicatus* Vockeroth
Aedes (*Ochlerotatus*) *increpitus* Dyar
Aedes (*Ochlerotatus*) *intrudens* Dyar
Aedes (*Finlaya*) *japonicus japonicus* (Theobald) (Jackson *et al.* 2016) (Updated distribution)
Aedes (*Ochlerotatus*) *mercurator* Dyar

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Aedes (*Ochlerotatus*) *melanimon* Dyar
Aedes (*Ochlerotatus*) *nevadensis* Chapman & Barr (Updated distribution, first formal record)
Aedes (*Ochlerotatus*) *nigripes* (Zetterstedt)
Aedes (*Ochlerotatus*) *pionips* Dyar
Aedes (*Ochlerotatus*) *provocans* (Walker)
Aedes (*Ochlerotatus*) *pullatus* (Coquillett)
Aedes (*Ochlerotatus*) *punctor* (Kirby)
Aedes (*Ochlerotatus*) *riparius* Dyar & Knab
Aedes (*Ochlerotatus*) *schizopinax* Dyar (Jackson *et al.* 2013)
Aedes (*Ochlerotatus*) *sierrensis* (Ludlow)
Aedes (*Ochlerotatus*) *spencerii spencerii* (Theobald) (Updated distribution)
Aedes (*Ochlerotatus*) *spencerii idahoensis* (Theobald) (Updated distribution)
Aedes (*Ochlerotatus*) *sticticus* (Meigen)
Aedes (*Ochlerotatus*) *togoi* (Theobald)
Aedes (*Aedes*) *vexans vexans* (Meigen)
Aedes (*Aedes*) *vexans nipponii* (Theobald) (Belton 2015) (First formal record)

Anopheles earlei Vargas
Anopheles freeborni Aitken
Anopheles punctipennis (Say) (Updated distribution)

Culex pipiens L.
Culex restuans Theobald (McCann and Belton 2015)
Culex tarsalis Coquillett (Updated distribution)
Culex territans Walker (Updated distribution)

Culiseta alaskaensis (Ludlow)
Culiseta impatiens (Walker)
Culiseta incidens (Thomson)
Culiseta inornata (Williston)
Culiseta minnesotae Barr
Culiseta morsitans (Theobald)
Culiseta particeps (Adams) (Jackson *et al.* 2013) (Updated distribution)
Coquillettidia perturbans (Walker) (Updated distribution)

Notes on new species records and distribution updates

Anopheles punctipennis (Say) is previously known from both Vancouver Island and the southern mainland of BC, but Darsie and Ward (2005) seem not to have recognized records of this species from Vancouver Island. Surveys by Stephen *et al.* (2006) found this species to be widely distributed on Vancouver Island, re-confirming its presence there.

Aedes aloponotum Dyar is known from the Fraser Valley and southern Vancouver Island. The distribution shown in Darsie and Ward (2005) seems to erroneously display the range of this species as extending up the Fraser Canyon and east to the interior of BC, possibly due to a mis-citation of Gjullin and Eddy (1972), who reported this species as occurring in the Fraser Valley. This may possibly be due to confusing the Fraser Valley with the Fraser Canyon. Additionally, the author has found this species from the outskirts of Whistler, extending the northern limits of its known range.

Aedes cinereus Meigen was reported from every part of BC by Belton (1983), but the distribution displayed by Darsie and Ward (2005) does not include Vancouver Island. Stephen *et al. (2006)* found *Ae. cinereus* in light traps on Vancouver Island, demonstrating that this species does occur there.

Aedes japonicus japonicus (Theobald) was first reported in BC from samples collected in Maple Ridge and Mission in 2014 (Jackson *et al.* 2016), and additional specimens have been collected by Sean McCann in Langley and by the author in Burnaby and Saanichton, with samples deposited in the Spencer Entomology Collection at the UBC Beaty Biodiversity Museum. This species seems to have become established in the Lower Mainland and southern Vancouver Island and may be spreading; if it is not present throughout these regions yet, it may soon become so. Whether or not it can become established in other parts of BC remains to be seen.

Aedes nevadensis Chapman and Barr was reported by Belton (1983) from larval collections made in Castlegar. However, this record does not seem to have been recognized by Darsie and Ward (2005), and its presence is recorded here to remove ambiguity. Belton made further collections of this species just outside Manning Park, and the author has collected them from Pemberton and from north of Princeton. The author's specimens have been deposited in the Spencer Entomology Collection at the UBC Beaty Biodiversity Museum. This species is likely found in dry areas of much of the Southern Interior of BC, although how far north its range extends is currently unknown.

Aedes schizopinax Dyar was first reported in BC from a collection made in the municipality of Sparwood, near the Alberta border (Jackson *et al.* 2013). An additional specimen, collected in Williams Lake by C. Phippen, along with records from Washington, suggest this species may exist in low numbers throughout the Interior of BC.

Aedes spencerii spencerii (Theobald) was previously believed to be present in BC only in the Peace River region (Belton 1983), with records from Kaslo of two specimens – one collected by HG Dyar and one by RP Currie (Dyar 1904) – considered dubious (Belton 1983). Examination of specimens in the Spencer Entomology Collection at the UBC Beaty Biodiversity Museum have revealed additional specimens of *Ae. spencerii spencerii* collected in the Southern Interior of BC, where it was previously believed only the *idahoensis* subspecies was found (Belton 1983). These two subspecies probably overlap in distribution throughout much of this region. I have also seen specimens in the Royal BC Museum collected from the Chilcotin.

Aedes togoi (Theobald) is thought to be an invasive species from Asia; however, there is evidence that this mosquito might be indigenous to rock pools along the coast of BC and adjacent Washington State (Sota *et al.* 2015).

Aedes vexans nipponii (Theobald) is a subspecies of *Ae. vexans* from east Asia that has recently been found in Ontario (Thielman and Hunter 2007). It is characterized by the presence of a median longitudinal stripe of pale scales on the abdominal tergites, which *Ae. vexans vexans* (Meighen) lacks (Tanaka *et al.* 1979). A specimen collected in Cawston by P. Belton distinctly possesses this attribute and has been deposited in the Spencer Entomology Collection at the UBC Beaty Biodiversity Museum.

Culex tarsalis Coquillett was previously thought to be found only in the southern half of mainland BC (Belton 1983; Wood *et al.* 1979). However, this vector of West Nile virus, Western equine encephalitis virus, and other viruses, has also recently been found in man-made sites throughout Vancouver Island (Stephen *et al.* 2006) and as far north as the Yukon (Peach 2018). It is likely to exist in suitable habitats throughout BC.

Culex territans Walker was reported as occurring across the south of BC by Belton (1983) but has also been found as far north as the Yukon (Belton and Belton 1990; Wood *et al.* 1979). Recent records extend its range to Vancouver Island (Stephen *et al.* 2006). These records imply that *Cx. territans* may be present throughout BC where suitable habitat exists.

Culiseta particeps (Adams) was first reported by (Jackson *et al.* 2013) from locations in Pitt Meadows and the Township of Langley. Additional specimens have also been found in Vancouver, including an adult female collected in 1918 that was found in a museum collection, and larvae that were found by the author in Prince Rupert. This species is likely to be found all along the coast of BC.

Coquillettidia perturbans (Walker) was previously known from suitable habitat throughout mainland southern BC (Belton 1983). Recent work by Poirier and Berry (2011) has revealed that this species is present as far north as Fort Nelson, and Stephen *et al.* (2006) found it throughout much of Vancouver Island, as well. These new records suggest it may be present in suitable habitat throughout most of BC, probably mirroring the distribution of host plants such as cattails (*Typha latifolia*) (Poirier and Berry 2011).

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